RESPONSES OF CAPTIVE COMMON EIDERS TO IMPLANTED SATELLITE TRANSMITTERS WITH PERCUTANEOUS ANTENNAS

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RESPONSES OF CAPTIVE COMMON EIDERS TO IMPLANTED SATELLITE TRANSMITTERS WITH PERCUTANEOUS ANTENNAS

A

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By

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Abstract

Implanted transmitters have been used for over a decade to track the migrations and habitat use of many sea duck species, but their effects remain largely unstudied. To address this, I assessed the physiological and behavioral responses and characterized the clinical responses of six Common Eiders implanted with a transmitter with a percutaneous antenna. To maintain a semi-natural feeding regime, I fed birds benthicly in a 4.9 m deep dive column. I collected blood, feces, mass, and video data prior to surgery to establish baselines and at staggered intervals for 3.5 months post-surgery to determine responses. All birds had some clinical complications, but most abated within 2 weeks of surgery. Mass increased in the first two weeks, but no trend was evident thereafter. Most biomarkers and dive performance metrics were altered at some point after surgery. While most biochemical values returned to baseline within weeks of surgery, a few remained deviated for longer. Additionally, dive speeds were slower for up to 3.5 months after implantation. Although it is uncertain how these changes would ultimately affect birds in the wild, effects on physiological condition and behavior seem likely in the first few weeks after surgery with longer-term effects also possible. Scientists should consider these responses and possible effects on the validity of PTT data when designing studies and analyzing information from implanted transmitters in sea ducks.

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DEDICATION

To my wife,

For her inspiration, humor, and love.

And my parents,

For their guidance and support.

"The important thing is not to stop questioning."

----Albert Einstein

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General Introduction:

Most sea ducks spend large portions of their lives at sea in areas difficult to access; thus the logistics and costs of studying them are often prohibitive. To learn more about demographics of animal populations, scientists developed platform transmitter terminals (PTTs) that transmit signals to polar orbiting satellites. While PTTs have proven beneficial for addressing key questions about sea duck ecology such as migration and habitat use, concerns remain as to how the devices affect the health of the carrier and consequently, the validity of provided data. Awareness of such effects is crucial, both when making inferences from PTT-provided information and in deciding the suitability of their use.

Radio tags were first used on wild birds in the 1960s, conveying physiological (Eliassen 1960; Lord et al. 1962) and location information (Southern 1964). Their utility has been limited in several ways including trackable distance, size, and method of attachment (see Korschgen et al. 1984; Strikwerda et al. 1986; Olsen et al. 1992; Benvenuti 1993; Kenward 2001). PTTs overcame the distance limitation by conveying data to orbiting satellites. Sizes have decreased substantially; early conventional (VHF) and satellite transmitters weighed 80 and 160 g, respectively (Southern 1964; Strikwerda et al. 1985), whereas today, conventional radios can weigh <1 g (WST2-12GL; Wildlife Track, Caldwell, ID, USA) and PTTs <10 g (PTT-100 Solar; Microwave Telemetry, Columbia, MD, USA). Attachment continues to be a challenge for many species of birds.

While the optimal method to assess the effects of transmitters would be to follow both a marked and control group in the wild, this is not possible for many sea duck species. Additionally, merely catching and banding control birds may bias results through capture myopathy (Dabbert and Powell 1993). To date, most studies have tested transmitter effect by focusing on overt ecological metrics such as survival and reproduction (Rotella et al. 1993; Garrettson and Rohwer 1998; Meyers et al. 1998; Iverson et al. 2006). While these metrics are important in determining transmitter efficacy, physiological markers are also useful (Kenward 2001; Schulz et al. 2005) and have been employed to determine responses of pigeons, passerines, and sea ducks to transmitters (Gessaman and Nagy 1998; Wells et al. 2003; Schulz et al. 2005; Mulcahy et al. 2007).

Effects associated with externally attached devices in waterfowl are numerous and include weight loss, abnormal behavior, decreased return rates, and increased preening (Perry 1981; Pietz et al. 1993; Robert et al. 2006). Diving birds are particularly susceptible due to complications associated with underwater drag (Wilson et al. 1986) and disruption of the insulating body feathers (Perry 1981). To minimize the effects of external transmitters, Korschgen et al. (1984; 1996) developed a technique to implant them into the abdominal cavity of waterfowl.

Responses of waterfowl to implanted transmitters have been less studied and more varied than external mounts. In initial trials of implanted radio transmitters (with internal antennas) in waterfowl, Korschgen et al. (1984) found 2 of 14 captive birds showed signs of infection but found no difference in time spent foraging, preening, or resting between implanted wild birds and controls. There was no substantial increase in post-release transmitter-attributable mortality in Canvasbacks (*Aythya valisineria*) implanted with transmitters with internal antennas (Olsen et al. 1992), and only 1% of Canada Geese (*Branta canadensis*) implanted with transmitters with percutaneous antennas died due to causes attributable to transmitters; 1-yr survival was similar to a control group (Hupp et al. 2006).

Most studies using implanted transmitters in sea ducks have reported relatively low bird mortality (e.g., Mulcahy and Esler 1999; Phillips et al. 2007); however, some found increased mortality (>5%) within the first few weeks of implantation (Rosenberg and Petrula 2000; Iverson et al. 2006). Both Esler et al. (2000) and Iverson et al. (2006) suggested a censor period post-surgery to reduce bias. Harlequin Ducks (*Histrionicus histrionicus*) with implanted transmitters lost more weight than banded controls in the first few weeks after surgery, though 1-yr mass change was not different (Esler et al. 2000) and only a few small changes were noted in 1-yr hematologies and biochemistries (Mulcahy et al. 2007). While studies have begun to address how sea ducks respond to transmitter implantation, a large gap still exists. Researchers do not have experimental data on biochemical or mass changes in the days to months after implant or inter-species differences in responses to implanted transmitters. Additionally, little is known as to how the implantation of these devices may affect diving, a critical part of sea duck ecology.

In this study I described clinical findings, and examined physiological and dive behavior responses of six captive Common Eiders (*Somateria mollissima*) to implanted transmitters with percutaneous antennas. Birds were housed outdoors, and all food was provided on the bottom of a 4.9 m deep enclosure to maintain a semi-natural feeding environment.

My specific objectives were to:

1. Describe clinical findings and determine mass, biochemical, and hematological responses of Common Eiders to surgery and subsequent carrying of an implanted PTT with percutaneous antenna.
2. Assess how surgery and carrying of an implanted PTT with percutaneous antenna affect the dive performance of Common Eiders.

In chapter 1, I address the first objective by examining changes in biochemistries. I also assess trends in mass after surgery and describe clinical conditions. In chapter 2, I address my second objective by testing for changes in dive behavior parameters.

Chapter 1:

Biochemical and Clinical Responses of Common Eiders to Implanted Satellite Transmitters[1]

Abstract: Small implantable platform transmitter terminals (PTTs) have been used widely to delineate populations and identify movement patterns of sea ducks, but effects of these devices remain relatively unexplored. To address this, we measured biomarker responses and characterized clinical responses of benthic foraging sea ducks implanted with a PTT and discuss how these responses could affect the validity of derived information. We trained 6 common eiders (*Somateria mollissima*) to dive to the bottom of a 4.9 m deep column for their food, allowed them to acclimate to these deeper dives, and implanted them with 38–47 g PTTs with percutaneous antennas. We collected behavioral, biochemical, and clinical data before surgery to establish baselines, and for 3.5 months post-surgery. The first feeding dive took place 22 hours post-surgery with 5 of 6 birds diving to the bottom within 35 hours of surgery. We found differences between baseline and ≥1 of 3 post-surgery periods (2–14, 20–28, and 55–105 days) in all primary biomarkers (creatine kinase, fecal glucocorticoid metabolites, albumin:globulin ratio, and packed cell volume) and 6 of 9 additional biomarkers (aspartate aminotransferase, heterophil:lymphocyte ratio,

[1]Latty, C.J., Hollmén, T.E., Petersen, M.R., Powell, A.N., and Andrews, R.D. 2008. Biochemical and clinical responses of common eiders to implanted satellite transmitters. Prepared for submission to Journal of Wildlife Management.

β_1-, β_2-, and γ-globulins, and albumin). Our findings show common eiders physiologically responded to PTTs for up to 2–3.5 months post-implantation, with the greatest response occurring within 14 days of implantation. These responses support the need for post-surgery data censor periods and should be given consideration when designing studies and analyzing information from PTTs in sea ducks.

1.2 INTRODUCTION

For over a decade, much of the information on distribution and migration of sea ducks has come from data provided by intra-peritoneal transmitters. These studies have provided important information for the conservation and management of many sea duck populations; however, responses of the carrier species to implanted devices have not been thoroughly characterized. Understanding of such responses is crucial, both when making inferences from derived information and in deciding the efficacy of transmitter use. Our study indirectly addresses a key assumption of any marking study: that neither the mark nor the marking process alters measured parameters during the time period of data collection (White and Garrott 1990).

The utility of PTTs has been primarily limited in two ways: size of the transmitter and attachment technique (Korschgen et al. 1984; Strikwerda et al. 1986; Benvenuti 1993). PTT weights have decreased substantially since first fitted on wild birds (Strikwerda et al. 1985), with attachment generally considered a greater problem for diving birds as they may be more susceptible to complications associated with underwater drag (Wilson et al. 1986) and disruption of the insulating body feathers

(Perry 1981). In an attempt to overcome harmful effects of external mounts, Korschgen et al. (1984; 1996) developed a surgical technique to implant transmitters into the abdominal cavity of waterfowl. Since its development the technique has been used to implant PTTs in $\geq$10 of 15 North American sea duck species (Petersen et al. 1995; Brodeur et al. 1999; Robert et al. 2000; Petersen and Flint 2002; Iverson et al. 2006; Mallory et al. 2006; Petersen et al. 2006; Phillips and Powell 2006; G. H. Olsen and M. C. Perry, U. S. Fish and Wildlife Service, unpublished data).

In studies comparing external and internal transmitters in waterfowl for use >2 months, implanting the device was preferable (Dzus and Clark 1996; Iverson et al. 2006); however, harmful effects have been described with both attachment techniques. Effects associated with externally attached devices in waterfowl are numerous and include weight loss, abnormal behavior, decreased return rates, and increased preening (Perry 1981; Pietz et al. 1993; Robert et al. 2006). Responses to implanted devices have been more varied. In initial trials of implanted radio transmitters (with internal antennas) in waterfowl, Korschgen et al. (1984) found 2 of 14 captive birds showed signs of infection and no difference in time spent foraging, preening, or resting between implanted wild birds and controls. There was no substantial increase in post-release transmitter-attributable mortality in canvasbacks (*Aythya valisineria*) implanted with transmitters with internal antennas (Olsen et al. 1992), and only 1% of Canada geese (*Branta canadensis*) implanted with radiotransmitters with percutaneous antennas died due to causes attributable to transmitters; 1-yr survival was similar to a control group (Hupp et al. 2006). In sea ducks, some studies found increased mortality (>5%) (Rosenberg and Petrula 2000; Iverson et al. 2006) and mass loss (Esler et al. 2000) within the first few weeks of implantation. However, most studies using implanted transmitters in sea ducks have had relatively low bird mortality (e.g., Mulcahy and Esler 1999; Phillips et al. 2007). Both Esler et al. (2000) and Iverson et al. (2006) suggested a censor period post-surgery to reduce bias.

Testing for changes in biomarkers has been found to be a useful technique to investigate transmitter effects (Kenward 2001), and was successfully used to determine the responses of pigeons and passerines to transmitters (Gessaman and Nagy 1998; Wells et al. 2003; Schulz et al. 2005). Additionally, Hollmén et al. (2001) and Wayland et al. (2002; 2003) found biomarkers a valuable method for assessing physiological condition in common eiders (*Somateria mollissima*).

Here we used primary (creatine kinase [CK], fecal glucocorticoid metabolites [FGMs], albumin:globulin ratio [A:G], and packed cell volume [PCV]) and secondary (lactate dehydrogenase [LDH], aspartate aminotransferase [AST], heterophil:lymphocyte ratio [H:L], serum protein fractions [albumin, α_1-, β_1-, β_2- and γ-globulins], and glucose) biomarkers to assess responses to implanted PTTs in common eiders. We chose primary parameters a priori based on specific physiological responses we wished to test: CK for muscle damage, FGMs for physiological stress, A:G for immunological response, and PCV for general health. We used secondary markers to further characterize responses and evaluate findings.

We used CK to evaluate muscle condition because the enzyme has been found to be a sensitive and specific indicator of muscle damage in birds (Lumeij et al.

1988a). CK has been used to assess damage caused by a variety of conditions including capture and handling, extreme exertion, and some medications (Bollinger et al. 1989; Dabbert and Powell 1993; Aktas et al. 1997; Guglielmo et al. 2001). We used AST and LDH to further assess the findings from CK because elevations of either combined with an elevation in CK provide additional evidence of muscle damage (Hochleithner 1994).

We used FGMs to evaluate stress response. Corticosterone is the primary stress hormone in birds (DeRoos 1961), and its metabolites are measurable in feces (Wasser et al. 2000; Ludders et al. 2001; Washburn et al. 2003). FGMs have been used to measure the stress response in another species of sea duck, the harlequin duck (*Histrionicus histrionicus*) (Nilsson 2004), and stress caused by transmitters (Wells et al. 2003). We used H:L to further assess stress responses (Gross and Siegel 1983) and compared findings with FGMs.

We employed A:G (Hochleithner 1994; Harris 2000) as a metric of inflammatory response. Albumin is the portion of blood proteins primarily comprised of non-immunological constituents, while globulins are mainly comprised of proteins involved in the response to both specific and non-specific immunological insults (Harris 2000). We used globulin fractions (Hochleithner 1994) to further characterize responses.

Because PCV is affected by a host of factors (Dein 1986; Campbell 1995), we used it as a general health index. Additionally, red blood cells provide significant oxygen storage for diving birds (Keijer and Butler 1982; Stephenson et al. 1989;

Butler 1991); therefore, decreased PCV may decrease an eider's ability to remain aerobic while submerged. We assessed for reductions in albumin and glucose to further evaluate overall health.

We chose common eider as our study species because they have been implanted with similar PTTs in field studies (Petersen and Flint 2002; Merkel et al. 2006), transmitter mass was within the recommended range (Kenward 2001), and their foraging depths (Guillemette et al. 2004) overlapped with the dive column depth. Additionally, their foraging ecology is broadly similar to those of threatened species for which an understanding of possible responses to implanted devices is listed as a management goal (K. Laing, U.S. Fish and Wildlife Service, unpublished report). Our primary objective was to determine and characterize short-term (≤3.5 months) physiological responses of common eiders to intra-abdominal PTTs. We addressed this objective by evaluating biomarkers in eiders before and after implantation of a 38-47 g PTT and describing observed clinical responses. We discuss the potential for these responses to affect the parameters that researchers might use PTTs to measure.

1.3 STUDY AREA

Common eider eggs salvaged from the Yukon-Kuskokwim Delta, Alaska were hatched at the Alaska SeaLife Center (ASLC) in Seward, Alaska in 2003. Birds were housed in an outdoor aviary with shallow pools (<1 m deep) prior to the experiment.

We conducted our experiment between September 2005 and March 2006 at the ASLC in a large, exposed outdoor aquarium, in which we constructed a dive column

(1.5 × 1.5 m wide and 4.9 m deep) with an attached terrestrial haul-out. We chose this time period because it was outside of molting and breeding seasons when biomarkers and mass are most likely to be affected by seasonality. We provided a soft roosting area and enclosed the aviary in nylon netting. The dive column was enclosed in plastic mesh and Plexiglas to allow free flow of saltwater and observation of diving.

1.4 METHODS

1.4.1 *EXPERIMENTAL DESIGN*

We used 6 eiders (3 male, 3 female; described herein as M1, M2, M3, F1, F2, and F3) in our experiment. Prior to the study, all birds were determined by veterinarians to be in good physiological health; all research was approved by the ASLC IACUC committee (05-005).

In the pre-experimental phase, we constructed a dive column, tested its usability, and dive-trained birds. Dive training consisted of providing food in a metal tray on the side of the dive column at progressively lower depths and was necessary because birds had not previously foraged at depths >1 m.

We began the experimental phase once birds were foraging for most of their food on the bottom of the dive tank. At the start of this phase, we allowed 53 days for acclimation to the deep diving regime. This acclimation period was essential for establishing accurate baselines because both diving waterfowl and mammals exposed to dive training morphologically and physiologically acclimate to increased demands (Stephenson et al. 1989; MacArthur et al. 2003). During the experimental phase, we collected baseline blood, feces, and mass; implanted birds with PTTs; and again collected blood, feces, and mass data at staggered intervals for 3.5 months post-surgery.

We divided blood collection dates into baseline (29 and 62 days pre-surgery and the day of surgery) and 4 post-surgery periods: combined post-surgery (pooled 2 – 105 days), A (2, 8, and 14 days), B (21 and 28 days), and C (56, 91, and 105 days). We also divided fecal collection dates into baseline (1, 2, and 3 days pre-surgery) and 4 post-surgery periods: combined post-surgery (pooled 3–104 days), A (3, 6, and 13 days), B (20 and 27 days), and C (55 and 104 days).

After assigning collection dates to specific periods, we determined the maximum or minimum (chosen a priori) values for each individual per period (maximums for CK, FGMs, AST, LDH, H:L, and globulins and minimums for A:G, PCV, albumin, and glucose). The use of either maximum or minimum was based on hypothesized direction of change and if change in one direction may be more deleterious than the other. For example, PCV can increase or decrease for a variety of reasons, but we hypothesized that: a) if a change were to occur post-surgery, it would most likely be reduced, and b) reduction of PCV in a diving bird would likely be more deleterious. We used maximum (minimum) per period for 3 main reasons: (1) comparing the maximum (minimum) of the 3 baseline values with the maximum (minimum) of the 2–3 values for each of the post-surgery periods minimized the number of pairwise tests performed, and (2) it allowed us to concentrate on the general trends during these periods and reduced small temporal differences in how individuals respond to a stimuli (such as the way individuals often have different time-lines for healing). For example, we thought it was more important to establish that all birds had lower A:G ratios during period A post-surgery than that M1, M2, and F3 had their lowest A:G ratios on day 2, and M3, F1, and F2 their lowest on day 8. Finally, (3) using all baseline values minimized the possibility that changes seen after surgery were simply due to normal deviations in a given chemistry. Had we used the average of the baselines, we would have ignored that some amount of variation is normal in biochemistry values. We characterized and described post-surgery clinical responses, but did not quantify or statistically evaluate them.

We fed birds a diet of Mazuri® sea duck pellets (Purina Mills, St. Louis, MO), blue mussels *(Mytilus edulis)*, and krill (*Euphausia superba*) prior to the experimental period, and a combination of Mazuri® sinking waterfowl pellets and blue mussels 4–5 times daily during the experimental period. We provided fresh water ad libitum to birds throughout the study.

1.4.2 *SATELLITE TRANSMITTERS*

Veterinarians implanted a PTT 100 (Microwave Telemetry, Columbia, MD) or 5130 PTT (HABIT Research, Victoria, British Columbia, Canada) when eiders were 29 months old using procedures similar to those developed by Korschgen et al. (1984; 1996). Approximate transmitter dimensions were 38–47 g, 70 x 35 x 15 mm, volume of 22.5 mL, with a 200 mm long, 1.7 mm diameter antenna. Surgery took place at the ASLC under aseptic conditions by veterinary surgeons experienced in PTT implantations. Briefly, they anesthetized birds using propofol (Machin and Caulkett 2000) and, in one case, isoflurane due to complications maintaining the I.V. They then placed a transmitter into the right abdominal air sac through a ventral, midline incision in the abdomen, with the antenna exiting the body dorsally and extending caudally from the bird. Two individual internal sutures were used to anchor a mesh housing (surrounding the transmitter) to the body wall, and one suture was used at the dorsal antenna exit site to secure the skin and muscle to a mesh collar attached to the base of the antenna inside the body wall. The abdominal incision was closed with 2 layers of monofilament absorbable suture. To maximize plumage integrity post-implant, feathers were not plucked at surgical sites

1.4.3 *BLOOD AND FECAL SAMPLING*

We captured birds in random order for each sampling event. We collected a 2–3 mL blood sample from the right jugular vein of each individual 62 and 29 days prior to surgery, the day of surgery, and at 2, 8, 14, 21, 28, 56, 91, and 105 days post-surgery between 0800 and 1200 Alaska Time. Handling sessions averaged 51 minutes (range 26–77 minutes) from first bird capture to last release. Time from capture to blood draw ranged from 0–8 minutes ($\bar{x}$ = 2 minutes). We did not feed birds on mornings of blood draws, though residual food may have been available from prior feedings. We analyzed blood samples the day of collection or froze serum at -80° C. To further characterize baseline variability, we also tested for differences using an extended baseline that included an additional 5 serum samples and hematology results obtained from each individual approximately every 2 months during the year prior to the start of the pre-experimental phase.

We collected fecal samples at 1, 2, and 3 days prior to surgery, and 3, 6, 13, 20, 27, 55, and 104 days after implant. We collected all samples with a wooden tongue depressor within 20 minutes of deposit and froze samples at -20° C within 3 hours of collection.

1.4.4 *BIOMARKER ANALYSES*

Unless stated otherwise we followed manufacturers' recommendations for analyzing chemistries. We used an i-STAT® PCA handheld analyzer and an EC8+ cartridge (Abbott Point-of-Care, East Windsor, NJ) to determine glucose and an IDEXX VetTest® chemistry analyzer and corresponding cartridge (IDEXX Laboratories, Westbrook, ME) to evaluate CK, AST, and LDH. For IDEXX tests, we diluted samples 1:2 serum / 0.9% saline, except a highly elevated CK and LDH sample, which we diluted to 1:8.

Total protein was analyzed at the Marshfield Clinic Laboratories (Marshfield, WI) using the biuret method. For protein fractions (pre-albumin, albumin, α_1-, α_2-, β_1-, β_2-, and γ-globulins) we used Serum Protein Electrophoresis (SPE) Kit (Beckman Coulter, Fullerton, CA) to separate and stain fractions and a Beckman Coulter densitometer to measure electrophoretic densities. We included pre-albumin and albumin in the albumin portion and all globulins in the globulin portion for A:G

(Harris 2000). Because the α_2-globulin fraction was not always discernable, we did not include it as a secondary parameter.

We determined white blood cell proportions by making ≥2 blood smears directly from the collection syringe using fresh untreated blood. After drying, we stained slides using a quick dip stain system (Jorgensen Laboratories, Loveland, CO). We then manually counted cells using a microscope. We determined the PCV ratio by centrifuging a capillary tube of heparinized blood for 3 minutes at 3500 rpm. We then used a Micro-Capillary Reader (IEC, Needham Heights, MA) to measure the ratio of cells to the total sample.

We used a ^{125}I Radioimmunoassay Kit (MP Biomedicals, Orangeburg, NY) for measuring FGMs, based on Nilsson's (2004) techniques for harlequin ducks. Pooled samples showed the assay accurately measured fecal metabolites (males: m = 1.01, R^2 = 0.994; females: m = 1.12, R^2 = 0.994). Serial dilutions of fecal extracts with steroid diluent yielded values parallel to the standard curve for both males and females. Mean intra-assay (between duplicates) coefficient of variation was 2.00%, and inter-assay coefficients of variation were 21.63 and 6.86% for controls with mean values of 85 and 621 ng/mL, respectively.

1.4.5 *MASS*

We weighed birds with a pesola spring scale 7 days prior to surgery. After implant, we recorded mass when birds stepped onto a waterproof bench scale (Rice Lake Weighing Systems, Rice Lake, WI) to drink from a freshwater bowl in the aviary. To ensure accuracy of measurements during subfreezing conditions, we placed a heat pad directly beneath the scale to keep temperatures within the manufacturer's recommended range. We recorded mass routinely after foraging bouts because birds generally drank from the water bowl post-feeding. Due to the opportunistic fashion of mass data collection, we did not necessarily obtain data for each individual at each weighing session.

1.4.6 *DATA ANALYSES*

We examined blood and fecal biomarkers using a multi-step approach in SAS® 9.1 (SAS Institute 2004) and considered tests significant at $\alpha = 0.05$. We tested for normality across the population of times and individuals for each parameter using a Shapiro-Wilk test, and applied log or rank transformations (Zimmerman 1996) as needed. We present means ± SD.

To determine appropriate baselines, we used mixed model, repeated measures analyses or Friedman tests to examine for linear changes during the acclimation period. For mixed models we included sex and time since surgery as effects and chose covariate model structure based on Akaike's Information Criterion corrected for small samples (AICc) (Littell et al. 1996). Only PCV ($F_{1, 11} = 10.76$, $P = 0.007$) changed in response to deeper diving during the pre-surgery acclimation period. Given this, we used the PCV values obtained the day of surgery as baseline. For all other parameters, we used each bird's maximum or minimum value from the 3 pre-surgery collection dates as baseline.

We examined primary parameters for differences between maximum or minimum baseline and pooled post-surgery values using a 1-tailed paired *t*-test. If a difference was found in this overall test, we conducted follow-up tests between baseline and periods A, B, and C; extended baseline and periods A, B, and C; and secondary parameters (all comparisons between maximum or minimum values) using a 1-tailed paired *t*-test. We controlled pairwise tests on primary markers (between baseline and overall and periods A, B, and C) with Holm-Bonferroni adjustments (Holm 1979). We considered secondary parameters and extended baselines supportive; therefore, no pairwise control was applied.

To establish if mass changed in response to the PTT, we used a repeated measures mixed model (Littell et al. 1996) to determine trends in percent mass change by examining 2 periods: 1–14 days and 1–119 days post-surgery. We included sex, number of feedings, and days since surgery as effects in our model.

1.5 RESULTS

1.5.1 *BEHAVIORAL/CLINICAL*

Surgeries took place on November 30, 2005, and averaged 19 minutes (range 11–29 minutes) from incision to closure. M3 suffered complications during surgery and required treatment for bradycardia. All birds showed signs of lethargy (drooping bill and wings) for 8–10 hours after surgery and some were seen shivering within 24 hours of surgery (minimum recorded temperature for Seward, Alaska on the 2 days post-surgery was -8° C). Lethargy was not noted on or after day 3, except for M1 (further described below). Though not quantified, we noted that birds spent much time preening surgical sites (especially the antenna exit site) for the first few weeks after surgery.

M3 was the first to dive to the bottom of the dive column 22 hours post-surgery. All but one bird dove to the bottom for food within 35 hours of surgery. The remaining bird (M1) showed signs of severe lethargy and appeared unable to climb an approximately 15 cm ramp between the water and dry aviary. This bird remained in the water until we manually removed it and placed it on the haul-out. Once removed, it had difficulty standing and holding up its bill. The bird was not performing foraging dives and we force-fed it to minimize the chance of permanent health problems. The bird's condition improved and it began performing foraging dives 50 hours after implant.

We found F2 had seepage of water into the plumage at the abdominal incision site, and 5 birds had wet plumage at the antenna exit site on day 2. The only bird without a wet antenna exit site on day 2 was M1, which had not yet begun diving. On day 8, we found no gross wetness of feathers at abdominal incision sites, but 4 birds had water intrusion into the plumage at the antenna exit site. Additionally, all birds had varying degrees of matting of contour and down feathers at the antenna exit site. We also found F2 had swelling without further signs of infection at the exit site on day 8. On day 14 we did not find wet plumage at either surgical site, but all birds had matting of contour and/or down feathers at antenna exit sites suggesting some loss of insulation and possibly waterproofing. We also noted several of the birds had been plucking feathers from around antenna exit sites exposing bare skin, with M3 having a 1.5 cm^2 plucked area. By 21 days post-surgery, all birds maintained waterproofing at both surgical sites, and those with bare skin a week prior were regrowing feathers.

M3 developed a limp after surgery, and on day 17 we found a wound on its foot that advanced to involve the webbing between the third and fourth toes and complete necrosis with disassociation of the phalanges of the fourth toe. A veterinarian amputated the affected toe 7 weeks post-surgery to minimize further health risks. Histopathology showed severe granulomatous necrotizing pododermatitis consistent with bacterial embolization and subsequent osteomyelitis. Because the etiology of this condition is uncertain and we cannot rule out the possibility that it was related to surgery, we included this individual in our analyses. The bird dove and successfully maintained condition despite being visibly irritated by the affected leg and sometimes not using it for propulsion during dives.

1.5.2 *BIOMARKERS*

All primary biomarkers deviated between baseline and the combined post-surgery period (Table 1). With follow-up pairwise tests, we found CK elevated in period A, FGMs elevated in period B, A:G decreased in period A, and PCV decreased during all periods (Table 1), with individuals generally following similar trends (Fig. 1.1).

For additional biomarkers of muscle condition, AST was elevated in both periods A and B, but there was no difference in LDH for any period (Table 1). Comparing the 3 post-surgery periods to the extended CK baseline, we found a difference only in period A (t_5 = -4.54, P = 0.003). The secondary stress parameter, H:L ratio, was elevated in periods A and B (Table 1).

For secondary inflammatory parameters, β_1- and γ-globulins were elevated in all periods, β_2-globulins were elevated in periods A and B, and α_1-globulins did not deviate in any period (Table 1). With extended A:G baseline, we found the same results, with only period A decreased (t_5 = 5.93, P = 0.001).

For other general health metrics, albumin was lower during period A and there were no differences in glucose during all periods (Table 1). With the extended PCV baseline, we found period A (t_5 = 2.21, P = 0.039) remained lower, but, unlike primary tests, we found no difference in periods B or C.

1.5.3 *MASS*

We recorded over 700 individual weights post-surgery with $\geq$1 measurement on 98 of the 119 post-surgery days. We removed gender from the model, but number of feedings was included ($F_{4, 20}$ = 144.87, $P \leq 0.001$). We found that birds overall gained mass (based on model solutions, an average of 128 g) between days 1 and 14 post-surgery (time [$F_{1, 123}$ = 96.01, $P \leq 0.001$]) (time^2 [$F_{1, 123}$ = 9.02, P =0.003]) (Fig. 1.2), but no change in the full 119 day term.

1.6 DISCUSSION

While the ultimate effect of responses on birds in the wild is uncertain, we found that captive common eiders physiologically responded to abdominally implanted PTTs with percutaneous antennas primarily for 2–14 days after surgery, but also for periods up to 2–3.5 months. In addition to changes in all primary and several secondary biomarkers, we observed general and individual clinical complications. Although our sample size appears sufficient to determine gross responses, large individual variation affected our ability to detect small-scale changes. One individual in particular (F3) often had values outside overall trends thereby increasing variation. Although our study did not enable inclusion of a control group due to size limitations of the aviary and dive column, we addressed possible non-PTT related seasonal variation within individuals by including extended baselines (an additional year prior to the start of the experiment) so that each bird had its own control period for the same season the previous year.

1.6.1 *BEHAVIORAL/CLINICAL*

Short-term (<15 day) increases in mortality are not uncommon in studies using transmitters (Cox and Afton 1998; Rosenberg and Petrula 2000) and mortality in sea ducks has been attributed to avian predators (Rosenberg and Petrula 2000; Iverson et al. 2006). Based on our observations, clinical responses such as lethargy, increased preening, and dive cessation may help explain increased mortality; eiders with these conditions could be at an increased risk of predation.

An important consideration in determining how implanted transmitters affect their carrier is to assess their impact on metabolic rates. While several of the observations we noted could affect energy use, plumage wetting probably has the greatest potential to increase energetic demands. For example, a small spot of plumage oiling and subsequent loss of waterproofing at 0° C increased the metabolic rate of mallards (*Anas platyrhynchos*) by approximately 30% (Hartung 1967). Although we observed shivering only in the first days post-surgery, thermoregulatory consequences may have continued longer. It is important to note that birds were not fed and therefore not diving prior to exams. Given the feather issues noted, it is possible waterproofing was reduced until matting abated. Additionally, wild eiders dive deeper than 5 m (Lovvorn et al. 2003; Heath et al. 2006; Mosbech et al. 2006), and increased water pressure may exacerbate plumage wetting.

1.6.2 *MASS*

Mass had an increasing trend during days 1–14, with no difference in the 107-d period. This shows that while birds were biochemically responding to surgery and the transmitter, condition was maintained. In contrast, wild harlequin ducks implanted with 15–17.5 g transmitters had a greater mass loss in the first few weeks after surgery than banded controls (Esler et al. 2000). The difference may be explained by disparities between captivity and the wild; in our study, birds were fed ad libitum in a feeding tray at the same depth daily and did not have to contend with the rigors or energetic demands of life in the wild. Had our birds performed the hundreds of daily foraging dives at varying depths that is normal for common eiders (Guillemette 2001), flown from area to area in search of prey, and dealt with at-sea stochastic conditions, mass may have responded differently. Additionally, we only assessed the overall 14-

day trend; therefore, some weight loss may have been present early in period A (as is evident in Figure 1.2). We did not conduct pairwise tests between baseline and post-surgery dates because data collection methods differed.

1.6.3 *BIOMARKERS*

Five birds had >100% increase in CK during period A, and four had levels associated with myopathy (Bollinger et al. 1989). CK was statistically elevated for ≤14 days after surgery (mostly on day 2; Appendix). However, elevated serum CK is a result of muscle damage; therefore, effects of damage may persist beyond the normalization of CK (Wobeser 1997). Also, the peak level and half-life of serum CK of budgerigars (*Melopsittacus undulates*) injected intra-muscularly with muscle extract was 4 and 7.7 hours, respectively (Itoh et al. 1993). Although not defined for common eiders or other waterfowl, if we assume moderately similar pharmacokinetics, the 48 hr post-surgery blood draw probably does not represent maximum post-surgery CK levels. While myopathy may reduce muscle performance and affect flight, diving, and movement, we caution that the use of muscle enzymes as a myopathy index has not been experimentally confirmed or evaluated for common eiders. Also, we cannot differentiate between CK elevation caused by the cutting of muscle during the operation from that of other sources, such as muscle affected by handling or hyperthermia.

Elevation of AST in period A supports suggested muscle damage. AST was elevated into period B (up to day 21; Appendix) consistent with the longer half-life of

AST compared to CK (Itoh et al. 1993), though this is longer than AST was elevated in the blood of racing pigeons induced with muscle damage (6 days) (Lumeij et al. 1988b). LDH was elevated on day 2 post-surgery (Appendix), but was not significantly different from baseline during period A. LDH is less specific for muscle damage (Hochleithner 1994) and has a shorter elimination half-life than either CK or AST (Itoh et al. 1993), which may have contributed to the lack of significant difference.

We used FGMs as our primary stress metric, though H:L has been shown as a valid measure of stress in birds (Gross and Siegel 1983). In Adelie penguins (*Pygoscelis adeliae*) with obvious injuries, elevations occurred in H:L but not serum corticosterone, leading Vleck et al. (2000) to suggest H:L may be a better index of persistent stress than blood corticosterone. We chose FGMs as a primary marker because immunological responses to surgery, the implanted device, and infection could affect H:L. This may explain why H:L was not in agreement with FGMs in period A.

Because A:G was depressed only during period A and secondary markers were not controlled for pairwise comparison, we advise caution when interpreting results of individual protein fractions beyond period A. Additionally, we caution that the constituents of the fractions have not been determined for eiders. That said, the elevation of β_1- for all periods, and β_2- for periods A and B suggests liver, kidney, or chronic inflammatory reactions during the entire study (Harris 2000). Elevation of γ-globulins (generally consisting of antibodies and complement) throughout post-surgery periods supports the conclusion of chronic inflammatory response and may relate to infection (Kaneko 1989). Although these elevations show implanted eiders were able to mount an immunological response, such responses have a metabolic cost. For example, collared doves (*Streptopelia decaocto*) immune challenged with sheep red blood cells had an 8.5% increase in basal metabolic rate compared to controls (Eraud et al. 2005), and the resting metabolic rate of great tits (*Parus major*) increased 4.5% in response to phytohaemagglutinin (Nilsson et al. 2007). While the magnitude of these increases was small, such changes could be biologically significant. We found no change in α_1-globulins for any period. Some proteins generally found in α-globulin portions are involved in acute phase reactions (Harris 2000). This lack of change suggests there was no elevation in these specific proteins. Further research is needed to understand the difference in responses among the different globulin fractions.

The tests between the extended PCV baseline and periods B and C indicated no difference. We advise caution in interpreting this finding because PCV increased in response to dive training; therefore the primary test (which used the day of surgery value as baseline) may be the better metric. In addition to a reflection of general health, reduced PCV could indicate lower oxygen storage capacity (Keijer and Butler 1982; Stephenson et al. 1989). This would likely reduce calculated aerobic dive limits (Hawkins et al. 2000) and therefore could impact foraging. We included albumin and glucose as increasingly robust metrics of health status. Reduced albumin in period A suggests a considerable effect in this period, while no change in glucose suggests this did not extend to malnutrition or septicemia (Woerpel and Rosskopf 1984). The disparity between mass gain and lower albumin and PCV during period A may be explained by birds being fed ad libitum and not being exposed to the rigors of life in the wild (increased number of dives, varied dive depth, searching for prey, flying, and stochastic conditions). Additionally, we only examined the overall 14-d trend and did not conduct pairwise tests because data collection methodology differed pre- and post-surgery. Based on model corrected mass change for days 1–14 post-surgery (Figure 1.2), some mass loss may have occurred during the first few days after surgery.

1.6.4 *CONCLUSIONS*

While the optimal method to determine if implanted PTTs affect derived information would be to follow a marked and control group throughout the year, such a study would not be possible for many sea duck species. Additionally, merely catching and banding control birds may cause capture myopathy, which could bias results (Dabbert and Powell 1993). The few studies that addressed implanted transmitter effects in sea ducks found no difference in survival among 4 types of attachment (intra-abdominal with and without percutaneous antennas, subcutaneous, and externally attached) (Iverson et al. 2006), that 1-yr survival and mass were similar to banded controls (Esler et al. 2000), and that AST, LDH, and uric acid were somewhat higher 1-yr post-implant (Mulcahy et al. 2007). Despite appearing normal just days after surgery and engaging in behaviors such as diving and feeding, we found physiologic responses to implanted PTTs mostly in the first few weeks after surgery, but also for up to 2–3.5

months post-implant. The range of clinical responses we observed implies variation in how individuals cope with surgery and carrying PTTs. Researchers using implanted PTTs need to consider both the suite of responses that occur and the variability of responses among individuals.

Although we cannot discriminate with certainty how the responses we found would ultimately affect wild common eiders, they do suggest health effects for at least the first few weeks after implantation. Further study is needed to determine longer-term responses, whether responses can be reduced by using smaller PTTs, if other sea duck species are similarly impacted, how responses to PTTs compare with those of other implanted devices (such as data loggers), and if responses vary with dive depth and/or season.

1.7 MANAGEMENT IMPLICATIONS

Based on our study, common eiders implanted with PTTs representing 1.9–2.6% body mass show physiological responses for up to 2–3.5 months post-surgery. Results here support the need for data censor periods, but additional studies on a wider range of physiological effects are needed before an appropriate censor period length can be determined. Until then, scientists should use the responses described here and findings of other applicable studies to assess the suitability of implanted transmitters with percutaneous antennas for their particular project, study species, and research questions.

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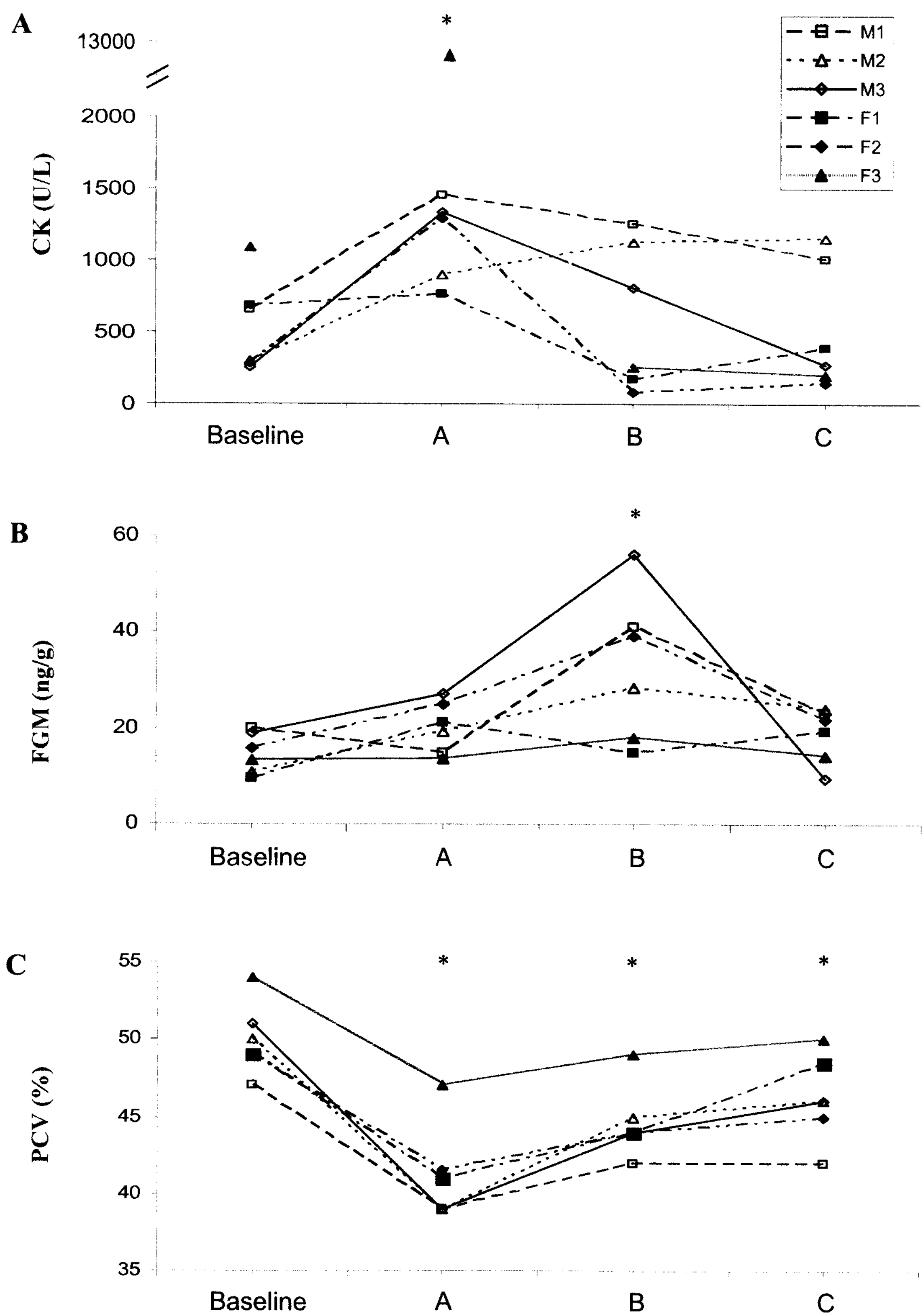
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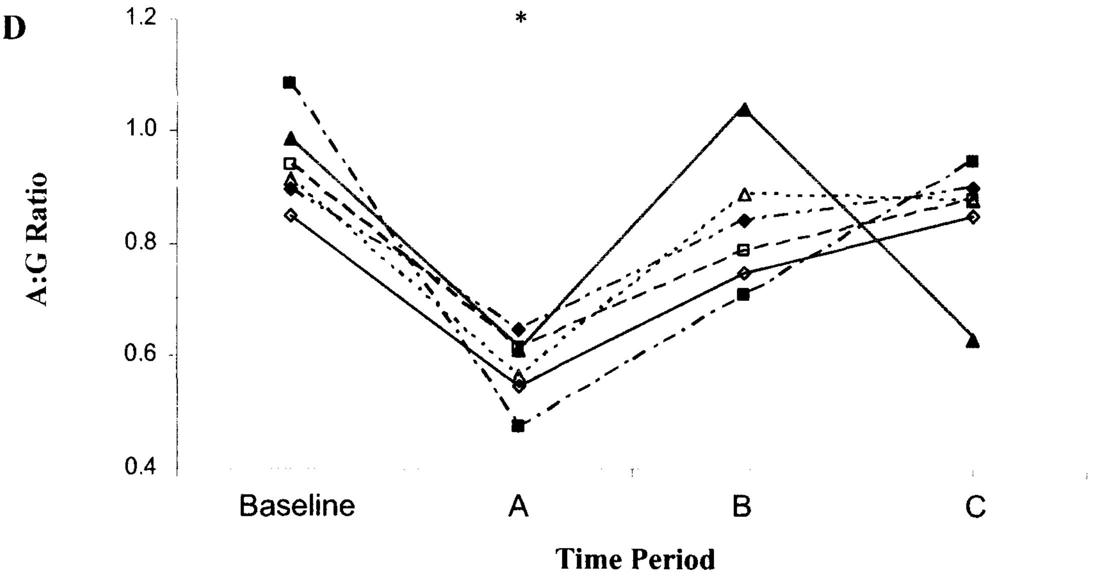

Figure 1.1 Maximum/minimum values of primary biomarkers through time. (A) max. creatine kinase (CK), (B) max. fecal glucocorticoid metabolites (FGMs), (C) min. packed cell volume (PCV), and (D) min. albumin:globulin ratio (A:G) of common eiders after abdominal implantation of satellite transmitters with percutaneous antennas (n = 6). We pooled data for 11 blood draws and 10 fecal collection days for each individual into 4 periods: baseline (blood: 2 mo pre-surgery through the day of surgery; fecal: 1-3 days pre-surgery), A (2-14 d), B (20-28 d), and C (55-105 d) and present the max. or min. values per period as described. Lines represent individuals through time. * indicates significant difference between baseline and appropriate term after adjustment with Holm-Bonferroni procedure.

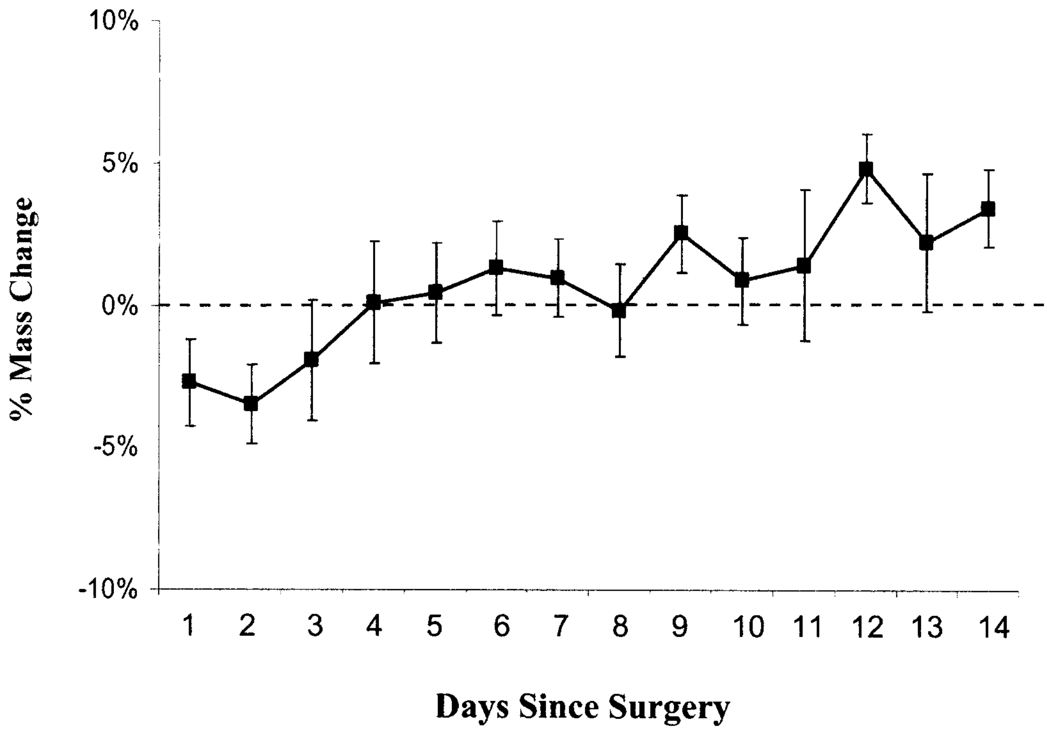

Figure 1.2 Mean % change in mass post-implant. Mean, adjusted % change ± SD in mass for days 1-14 after abdominal implantation of a satellite transmitter for 6 common eiders. To correct for number of feedings, we adjusted mass based on repeated measures mixed model solutions. To determine daily % change, we compared corrected post-surgery mass to corrected mass 7 days prior to implant. For days 1 and 4, n = 4; for days 3 and 12, n = 5; for others, n = 6.

Table 1 Summary of biomarker responses. Biomarker responses of common eiders implanted with 38–47 g satellite transmitters with percutaneous antennas. Differences in markers were determined using 1-tailed paired *t*-tests (df = 5). We controlled pairwise tests on primary markers (CK, FGM, A:G, and PCV) with a Holm-Bonferroni procedure.

Parameter	Term	Mean Values ± SD	% Change from Baselines (Mean ± SD and Range)	t	P
Max. CK	Baseline	544 ± 327			
(U/L)	Post (2–107 d)	3092 ± 4644	374 ± 365 (13 to 1053)	-5.08	0.002*
	A (2–14 d)	3049 ± 4667	359 ± 371 (13 to 1053)	-4.54	0.003*
	B (21– 28 d)	617 ± 512	60 ± 158 (-77 to 274)	0.14	0.553
	C (2–3.5 mo)	527 ± 439	2 ± 134 (-82 to 283)	0.47	0.671
Max.	Baseline	15 ± 4.36			
FGM	Post (2–105 d)	34 ± 14.21	130 ± 55 (37 to 196)	-6.64	≤ 0.001*
(ng/g)	A (2–14 d)	20 ± 5.39	48 ± 55 (-25 to 128)	-1.98	0.052
	B (21– 28 d)	33 ± 15.54	119 ± 62 (37 to 196)	-6.50	≤ 0.001*
	C (2–3.5 mo)	19 ± 5.68	41 ± 66 (-50 to 125)	-1.49	0.093
Min. A:G	Baseline	0.95 ± 0.08			
(ratio)	Post (2–107 d)	0.58 ± 0.06	-38 ± 9.43 (-56 to -28)	7.26	≤ 0.001*
	A (2–14 d)	0.95 ± 0.06	-38 ± 9.43 (-56 to -28)	7.26	≤ 0.001*
	B (21– 28 d)	0.84 ± 0.11	-11 ± 13.7 (-35 to 5)	1.81	0.065
	C (2–3.5 mo)	0.85 ± 0.11	-10 ± 13.8 (-36 to 0)	1.74	0.071
Min. PCV	Baseline	50 ± 2.37			
(% cells)	Post (2–107 d)	41 ± 3.11	-18 ± 4.07 (-24 to -13)	10.60	≤ 0.001*
	A (2–14 d)	41 ± 3.11	-18 ± 4.07 (-24 to -13)	10.60	≤ 0.001*
	B (21– 28 d)	45 ± 2.34	-11 ± 1.56 (-14 to -9)	16.00	≤ 0.001*
	C (2–3.5 mo)	46 ± 2.79	-8 ± 3.40 (-11 to -1)	5.51	0.001*
Max. α_1-	Baseline	1.03 ± 0.05			
globulins	A (2–14 d)	1.01 ± 0.08	-1 ± 7 (-10 to 10)	0.49	0.677
(g/dL)	B (21– 28 d)	0.96 ± 0.08	-6 ± 11 (-22 to 10)	1.43	0.894
	C (2–3.5 mo)	1.01 ± 0.6	-1 ± 8 (-9 to 13)	0.39	0.644

Table 1 cont.

Max. β1-globulins (g/dL)	Baseline	0.27 ± 0.03			
	A (2–14 d)	0.37 ± 0.05	40 ± 18 (9 to 59)	-5.76	0.001
	B (21–28 d)	0.31 ± 0.03	15 ± 11 (0 to 33)	-3.67	0.007
	C (2–3.5 mo)	0.31 ± 0.05	17 ± 10 (11 to 36)	-4.80	0.002
Max. β2-globulins (g/dL)	Baseline	0.62 ± 0.14			
	A (2–14 d)	1.15 ± 0.21	91 ± 40 (54 to 158)	-7.69	≤ 0.001
	B (21–28 d)	0.93 ± 0.16	55 ± 34 (25 to 115)	-5.07	0.002
	C (2–3.5 mo)	0.83 ± 0.30	39 ± 53 (-17 to 131)	-1.86	0.061
Max. γ-globulins (g/dL)	Baseline	0.17 ± 0.03			
	A (2–14 d)	0.23 ± 0.08	34 ± 36 (-9 to 84)	-2.23	0.036
	B (21–28 d)	0.26 ± 0.09	49 ± 37 (-2 to 97)	-3.51	0.009
	C (2–3.5 mo)	0.19 ± 0.03	15 ± 18 (-3 to 45)	-2.14	0.043
Max. H:L (ratio)	Baseline	1.07 ± 0.74			
	A (2–14 d)	3.14 ± 2.08	269 ± 294 (47 to 859)	-4.32	0.004
	B (21–28 d)	1.57 ± 0.63	122 ± 167 (-20 to 428)	-2.01	0.050
	C (2–3.5 mo)	0.99 ± 0.69	116 ± 307 (-58 to 716)	-0.14	0.447
Min. Albumin (g/dL)	Baseline	1.85 ± 0.23			
	A (2–14 d)	1.34 ± 0.21	-27 ± 11 (-37 to -14)	5.41	0.001
	B (21–28 d)	1.96 ± 0.26	7 ± 16 (-14 to 26)	-0.90	0.795
	C (2–3.5 mo)	1.92 ± 0.25	4 ± 8 (-9 to 17)	-1.08	0.835
Min. Glucose (mg/dL)	Baseline	199 ± 27			
	A (2–14 d)	189 ± 21	-4 ± 17 (-23 to 21)	0.73	0.249
	B (21–28 d)	203 ± 15	4 ± 17 (-21 to 24)	-0.30	0.612
	C (2–3.5 mo)	202 ± 22	3 ± 20 (-26 to 31)	-0.18	0.568
Max. AST (U/L)	Baseline	1.17 ± 2.86			
	A (2–14 d)	21.67 ± 18.72	[a]	-5.3	0.002
	B (21–28 d)	26.33 ± 19.33	[a]	-2.53	0.026
	C (2–3.5 mo)	0 ± 0	[a]	1.00	0.818

Table 1 cont.

Max. LDH	Baseline	972 ± 351			
(U/L)	A (2–14 d)	1356 ± 1517	47 ± 148 (-71 to 341)	0.77	0.762
	B (21– 28 d)	596 ± 101	-34 ± 18 (-62 to -16)	4.44	0.997
	C (2–3.5 mo)	796 ± 217	-11 ± 29 (-68 to 10)	0.85	0.783

[a] No percent change given for AST because 5 of 6 baseline values were equal to 0.

[*] Indicates statistical significance after adjustment with Holm-Bonferroni.

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Chapter 2:

Abdominally Implanted Transmitters with Percutaneous Antennas Affect the Dive Performance of Common Eiders[1]

2.1 *Abstract:* Implanted transmitters have become an important tool for studying sea duck ecology, but their effects remain largely undocumented. We assessed how abdominally implanted transmitters with percutaneous antennas affect the vertical dive speeds, stroke frequencies, and dive duration of captive Common Eiders (*Somateria mollissima*). We recorded video of six birds diving 4.9 m prior to surgery to establish baselines, implanted birds with 38–47 g platform transmitter terminals, and then recorded diving at staggered intervals for 3.5 months post-surgery to determine effects. Descent speeds were 16–25% slower post-surgery and remained below baselines at the end of the study. Ascent speeds were 17–44% slower for up to 2 months after implantation. Dive durations were longer than baseline until day 22. Foot stroke frequencies while foraging on the bottom were slower for most measurement days between 15–107 days post-surgery. If birds that rely on benthic invertebrates for sustenance are inferior divers after being implanted with a satellite transmitter, some repercussions are inevitable. Researchers considering use of implanted transmitters with percutaneous antennas should be mindful of these

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effects and the likelihood of concomitant alterations in foraging success, migratory behavior, and possibly even survival compared with unmarked conspecifics.

2.2 INTRODUCTION

Implanted radio transmitters have revolutionized scientists' ability to determine the movement patterns of sea ducks. Platform transmitter terminals (PTTs) specifically have led to the discovery of migration corridors and wintering areas, and allowed researchers to better understand migration timing and habitats (Petersen et al. 1995; Brodeur et al. 2002; Petersen et al. 2003; Petersen et al. 2006; Phillips et al. 2006). Regardless, concerns about health effects and the validity of information provided by these devices remain. Determining effects on measured parameters has been difficult due to the often harsh and remote environments sea ducks inhabit. To fill this gap, researchers have generally relied on ecological and physiological parameters to determine whether there are detrimental effects of manipulation and transmitter implantation. Although diving is central to sea duck ecology, no studies have addressed if and how implanted transmitters with percutaneous antennas affect sea duck dive performance.

Most of the research on how transmitters and other devices affect diving has been performed on penguins using external attachment. These studies found numerous effects including increased cost of transport, longer foraging trip durations, lower foraging success, and reduced swim speeds (Wilson et al. 1986; Croll et al.

1991; Culik and Wilson 1991). In some cases, effects were attributed to increased drag (Croll et al. 1991; Croll et al. 1996; Ropert-Coudert et al. 2000). The few studies on diving waterfowl found external attachment led to abnormal dive behavior and reduced foraging (Woakes and Butler 1975; Perry 1981; Robert et al. 2006).

In an attempt to reduce the effects of external mounts, Korschgen et al. (1984; 1996) developed a technique to implant the transmitter into the abdominal cavity of waterfowl. This technique has since been used with most sea duck species (e.g., Petersen et al. 1995; Petersen et al. 2006; Phillips et al. 2006). Although implantation should reduce external mount complications, effects of surgery and carrying the device internally are still possible.

Few studies have investigated how implanted devices affect the diving of birds. In Macaroni Penguins (*Eudyptes chrysolophus*), 20 g dataloggers had no effect on foraging trip duration (Green et al. 2004). Adelie Penguins (*Pygoscelis adeliae*) implanted with dataloggers swam more efficiently at slow speeds than controls, though the sample size was small (n = 2) and implanted birds may have been habituated (Culik and Wilson 1991). Although no studies have measured dive-related effects for sea ducks with implanted devices, some used reproduction and return rates (Guillemette et al. 2002), presumed or known mortality (Rosenberg and Petrula 2000; Iverson et al. 2006), recapture rates and mass change (Esler et al. 2000), and biochemistry and hematological responses (Mulcahy et al. 2007) to assess device effect.

While the optimal method to determine whether implanted transmitters affect derived information would be to follow a marked and control group throughout the year, such a study would not be possible for many sea duck species. Additionally, merely catching and banding control birds may cause capture myopathy (Dabbert and Powell 1993), which could bias results. Because changes in diving could affect the parameters scientists use implanted telemetry to measure, we assessed device effect by measuring changes in dive performance. To do this we recorded video of six Common Eiders diving before surgery to establish baselines, implanted birds with PTTs with percutaneous antennas, and then recorded additional video at staggered intervals for 3.5 months.

We chose Common Eider as our study species because they have been implanted with similar PTTs in field studies (Petersen and Flint 2002), transmitter-to-body mass ratio was within the recommended range (Kenward 2001), and foraging depths in the wild (Guillemette et al. 2004) were consistent with the depth of our dive column. Additionally, their ecology is broadly similar to that of threatened species for which an understanding of transmitter effects is listed as a management goal (K. Laing, U.S. Fish and Wildlife Service, unpublished report). Common Eiders feed on a variety of benthic prey including mussels, clams, urchins, and crabs (reviewed in: Goudie et al. 2000; Leopold et al. 2001). Their diving is constrained by factors such as available oxygen stores, buoyancy, and drag (Stephenson et al. 1989a; Lovvorn et al. 1991; Hawkins et al. 2000). Common Eiders overcome these forces using coordinated feet and wing strokes for propulsion during descent, and foot strokes only while foraging on the bottom (Heath et al. 2006). Ascent is passive, relying on positive buoyancy to return birds to the surface.

We hypothesized that due to the invasiveness of surgery, and possibly the size and density of the implanted device, vertical travel speeds would be affected by the implantation of a PTT into the abdominal cavity of Common Eiders. We also assessed the effect of transmitter implant and internal presence of the device on dive duration, foot/wing stroke frequency while diving, and foot stroke frequency on the bottom.

2.3 METHODS

Common eider eggs salvaged from the Yukon-Kuskokwim Delta, Alaska were hatched at the Alaska SeaLife Center (ASLC) in Seward, Alaska in 2003. Birds (three males and three females; pre-surgery mass 1800–2040 g) were housed in an outdoor aviary with shallow pools (<1 m deep) prior to the experiment. We fed birds Mazuri® sea duck pellets (Purina Mills, St. Louis, MO, USA), blue mussels *(Mytilus edulis)*, and krill (*Euphasia suberba*) prior to the experimental period and Mazuri® sinking waterfowl pellets and blue mussels during the experimental phase.

We conducted our study at the ASLC in an outdoor seawater aquarium, in which we constructed a dive column (1.5 × 1.5 m wide and 4.9 m deep) with an attached terrestrial haul-out. We enclosed the aviary with nylon netting and the dive column with plastic mesh and Plexiglas. We covered the aviary floor with Nomad matting (Safety-Walk 1500, 3M, St. Paul, MN, USA) to minimize injury risk to birds. We passed food through a PVC pipe onto an acrylic feeding tray on the floor of the dive column four to five times daily, thus allowing only benthic foraging. Prior to collecting baseline data, we trained birds to dive to deeper depths. Dive training consisted of providing food in a metal tray on the side of the dive column at progressively lower depths and was necessary because birds had not previously foraged at depths >1 m.

We began the experiment in September 2005 once birds were foraging for most of their food on the bottom of the dive column. At the start we allowed 53 days for acclimation to deeper diving. This was essential because diving waterfowl exposed to dive training acclimate to increased demands (Stephenson et al. 1989b). Veterinarians deemed birds were in good physiological health prior to the study. This study was approved by the ASLC IACUC committee (05-005).

Due to our small sample size, we established *a priori* two types of parameters of interest: primary (descent and ascent rates and wing/foot stroke frequency during descent) and exploratory (dive duration and foot stroke frequency on bottom). The functional difference between these was that we controlled primary parameter statistical tests for multiple comparisons, but did not for exploratory parameter statistical tests. The rationale for this was simple: minimizing controlled tests maximized our ability to detect change.

One male (hereafter referred to as bird A) developed necrosis of a toe and surrounding webbing after implant. Histopathology showed severe granulomatous necrotizing pododermatitis consistent with bacterial embolization and subsequent osteomyelitis. Because the etiology of this condition was uncertain and we could not rule out a surgical link, we included this bird in primary tests, but also present primary parameters excluding this bird in the exploratory section.

2.3.1 *SATELLITE TRANSMITTERS*

Birds were implanted when they were 29 months old with either a PTT 100 (Microwave Telemetry, Columbia, MD, USA) or a 5130 PTT (HABIT Research, Victoria, British Columbia, Canada) using procedures similar to Korschgen et al. (1984; 1996). Approximate transmitter dimensions were 38–47 g, 70 x 35 x 15 mm, volume of 22.5 mL, with a 200 mm long, 1.7 mm diameter antenna. Surgery took place at the ASLC under aseptic conditions by veterinary surgeons experienced in the technique. Briefly, they anesthetized birds using propofol (Machin and Caulkett 2000) and, in one case, isoflurane due to complications maintaining the I.V. A transmitter was placed into the right abdominal air sac through a ventral, midline incision in the abdomen, with the antenna exiting the body dorsally and extending caudally from the bird. Two individual internal sutures were used to anchor a mesh housing (surrounding the transmitter) to the body wall, and one suture was used at the dorsal antenna exit site to secure the skin and muscle to a mesh collar attached to the base of the antenna inside the body wall. The abdominal incision was closed with two layers of monofilament absorbable suture. To maximize plumage integrity post-implant, feathers were not plucked at surgical sites.

2.3.2 *VIDEO RECORDING AND ANALYSIS*

We examined over 850 dives using five to seven color digital video recording devices: three on horizontal planes, one recording foraging on bottom, and one to three capturing surface activities (Figure 2.1). We captured footage at 29.97 frames s^{-1} in long-play format on mini-DV tapes (Panasonic AY-DVM63PQ, Panasonic Corporation, Secaucus, NJ, USA). We transferred video from tapes to computer files in uncompressed Audio Video Interleave format and analyzed data in Adobe Premiere Pro 2.0 (Adobe Systems, San Jose, CA, USA). We recorded approximately 10 hrs of baseline video 2–4 days prior to surgery and 5–6 hrs of video on days 1, 2, 3, 4, 9, 15, 22, 29, 58, and 107 post-implant. Because some birds performed very few dives in the days directly after surgery, and all birds did not begin diving the same day, we combined days 1–4 in analyses. Additionally, we used footage from day 14 to augment day 15 video due to camera failure during a recording session.

We measured dive speeds and descent foot/wing stroke frequency between ~1.5–3.5 m of water. We began counts the first frame a bird passed a marker on a horizontal plane with the camera (Figure 2.1; marker A) and continued until the bird passed a similar marker for the next camera (Figure 2.1; marker B). We separated foot/wing strokes into four stages with similar durations (feet retracted, kick thrust, feet in glide stage with wings on downstroke, and feet in glide stage with wings on upstroke) based on descriptions of Heath et al. (2006) and the behavior of birds in this study. We calculated foot strokes on the bottom starting the frame a bird began pecking for food on the bottom and ended when foot strokes ceased and the bird began passively rising from bottom. Foot strokes on the bottom and foot/wing strokes during descent were divided by time to obtain frequencies. For dive duration, we considered a dive to start when a bird's bill first entered the water and stop when any part of the bird arose from the water.

We only analyzed dives where food was present and we saw birds feeding. We excluded parts of dives from analyses for a variety of reasons; thus, the number of dive portions analyzed varied greatly. Over the entire time series, individual descent rate, ascent rate, and foot/wing stroke frequency medians for a given day were generated from 6–27, 4–28, and 4–26 dives, respectively. This large range was due to variance in individual behavior. We excluded descent rates and foot/wing stroke frequencies if birds interacted with other birds, searched for food, or paused during descent. We excluded ascent rates if birds searched for food, handled food items, or interacted with other birds during ascent. We excluded dive durations if birds interacted with other birds at any point during the dive. We only examined foot stroke frequency on the bottom when three or fewer birds were present because as the number of birds foraging increased, discerning an individual's foot strokes became more difficult and interactions among birds more common. We also excluded sequences for foot stroke frequency on the bottom if birds rose >1 m from bottom while foraging.

Coordinated foot/wing strokes were the only mode of descent described by Heath et al. (2006) in wild Common Eiders. However, here, in addition to primarily relying on coordinated strokes, birds sometimes used foot-only propulsion during descent. Based on the hypothesis that surgery or carrying the transmitter may alter behavior (Perry 1981; Culik and Wilson 1991), we used all methods of propulsion in our primary descent rate calculations. We also present descent rates for dives containing only coordinated foot/wing strokes as a separate analysis in the exploratory section.

2.3.3 *STATISTICAL ANALYSES*

We analyzed data using SAS® 9.1 (SAS Institute 2004) and considered tests significant at $\alpha = 0.05$. We assessed normality with a Shapiro-Wilk test and applied log transformations to ascent rates and dive durations. We used a repeated measures mixed model to analyze primary parameters (Littell et al. 1996). If an overall test was significant, we used paired *t*-tests to check for differences between baseline and post-surgery periods. We controlled primary parameter tests with a Holm-Bonferroni procedure (Holm 1979). For exploratory parameters, we used paired *t*-tests to determine differences between baseline and post-surgery days. The purpose of this project was to assess deleterious effects; therefore, we used one-tailed tests to assess if descent and ascent rates were slower post-surgery. We used two-tailed tests for all other parameters. Means are presented ± SD.

2.4 RESULTS

2.4.1 *PRIMARY PARAMETERS*

Descent and ascent rates varied across respective time series ($F_{7,35} = 5.18$, $P = <0.001$, $F_{7,35} = 10.59$, $P = <0.001$). We found no difference for foot/wing stroke frequency. With pairwise tests, descent rates on all but day 15, and ascent rates on days 1–4, 9, 22, 29, and 58 were slower than baselines (Table 2.1). Median descent and ascent rates decreased an average of 16–25% and 17–44%, respectively, compared to baselines (Figure 2.2).

2.4.2 *EXPLORATORY PARAMETERS*

Coordinated foot/wing stroke descent rates were slower than baselines except for day 15 (Table 2.2) with average change for significant dates 13–20% below baseline (Figure 2.3). Dive durations were longer on days 1–4, 9, and 15 (Table 2.2) with average median dive duration increasing 13–19% for those days (Figure 2.4). Foot stroke frequency while foraging on the bottom became lower on day 15 and remained lower on days 22, 58, and 107 (Table 2.2) with the average ranging 4–7% below baselines for those days (Figure 2.5). Excluding bird A, median ascent was slower for all days and median descent was slower for all but day 15 (Table 2.3). We found no difference between baseline and post-surgery dates for foot/wing stroke frequencies during descent when bird A was excluded (Table 2.3).

2.5 DISCUSSION

For seabirds, dive speeds are generally conserved within relatively narrow ranges (Watanuki et al. 2005; Heath et al. 2006; Watanuki et al. 2006). Birds do this to maintain muscle efficiency (Lovvorn 2001; Watanuki et al. 2005) and minimize drag (Lovvorn et al. 1999) while maximizing travel speed, thereby minimizing their cost of transport (COT) (Lovvorn et al. 2004). Here we found the travel speeds of Common Eiders were slower after the implantation of a transmitter with percutaneous antenna.

As descent speeds slow, COT, a key metric of dive efficiency (measured as power input per distance traveled) (Wilson et al. 2004), will likely increase (Culik et al. 1993, 1994). This is because at speeds slower than ~1.25 m s^{-1}, drag (and power to overcome drag) changes slightly, but as speeds increase beyond ~1.25 m s^{-1}, drag and power increase in a rapid, non-linear fashion with speed. Therefore travel is most efficient (lowest COT) when drag and speed are balanced.

In addition to possible changes in COT, increased time spent descending could contribute to elevated costs of diving. The descent phase of dives is 2.9–3.3 times more costly than time spent at the bottom for diving ducks (Lovvorn et al. 1991). This suggests that even small alterations to descent duration could appreciably increase the overall power requirements for a dive and may limit foraging time on the bottom.

The dive speeds we found for captive Common Eiders diving 4.9 m were slower than were found for wild Common Eiders diving 11.3 m (Heath et al. 2006, 2007). This may be explained by differences in depth or between subspecies, constraints of the dive column, or wild versus captive birds. Regardless, assessing how implanted PTTs may affect deeper dives is important because for deeper depths, bottom time was inversely related to travel time (Heath et al. 2007). Using our descent/ascent rates, eiders diving 11.3 m would have a median travel time of 22 sec pre-transmitter implant, 33 sec 1–4 days post-implant, and 27 sec 107 days post-implant. Using speeds in the range found for wild Common Eiders diving at this depth (1.25 m s^{-1}) (Heath et al. 2006; Heath et al. 2007) and applying our relative changes, travel times would be 18 sec without the PTT, 25 sec 1–4 days after surgery, and 23 sec 107 days after surgery. Comparing these times with calculated aerobic dive limits (cADLs) for Common Eiders (51 sec) (Hawkins et al. 2000), implanted birds would have 19–37% less time to aerobically forage 1–4 days after implant and 14–21% less on day 107 post-implant. Additionally, reduced hematocrit, possibly reduced myoglobin (based on enzyme suggested myopathy), and higher COT after transmitter implantation may reduce ADLs. While extrapolation provides a valuable example of how deeper dives may be affected, we caution that dive performance effects may vary with depth.

2.5.1 *EXPLANATION OF EFFECTS*

2.5.1.1 BIOMECHANICAL

Buoyancy is a primary factor birds must overcome for shallow dives (Stephenson et al. 1989a; Lovvorn and Jones 1991b; Stephenson 1994), and decreased buoyancy is a potential cause of ascent and foot stroke frequency changes seen here. Based on a 22.5 mL, 42.1 g PTT, buoyant force would be reduced ~ 0.41 N. Using a mass specific buoyancy model (Lovvorn and Jones 1991a), this represents a 2.8–3.6% reduction from pre-surgery levels. This compares to a 20% decrease in Tufted Duck (*Aythya fuligula*) buoyancy due to normal plumage air loss during 1.5 m dives (Stephenson 1994). Additionally, this assumes birds do not adjust their diving lung volumes to ameliorate the effect of the transmitter. While the calculated decrease in buoyancy due to the transmitter appears small, implantation could affect buoyancy in other ways such as air sac rupture during surgery and plumage wetting at surgical sites. We also note that changing lipid stores could have affected buoyancy (Lovvorn and Jones 1991b). Because we measured parameters during the winter (November to March), we do not expect fat to lean body mass changed much, but cannot be sure since this was not tested.

Balance in the water has been suggested to alter diving in birds fitted with external devices (Healy et al. 2004) and may have affected birds here. Though not quantified, we noted birds after surgery ascended differently. Pre-surgery, most birds ascended in a tight, semi-circular fashion with body positions often nearing vertical, but after surgery birds tended to ascend in a wider circle with bodies more horizontal.

Although drag is reduced by implanting transmitters compared to using an external mount, percutaneous antennas still produce drag. If antenna drag was substantial, it could explain changes in both descent and ascent. However, drag produced by a 200 X 1 mm antenna on Magellanic Penguins (*Spheniscus magellanicus*) traveling at slow speeds (<1.25 m s^{-1}) was minimal and caused little or no increase in COT (Wilson et al. 2004).

2.5.1.2 PHYSIOLOGICAL

Mechanical force changes alone do not appear to fully explain slower travel speeds after PTT implantation. Based on elevated muscle enzymes in the blood, myopathy may have led to lower propulsive force generation per stroke post-surgery.

Four of our birds had short-term (≤14 day) CK levels associated with myopathy (Bollinger et al. 1989). These birds also had slower median swim speeds than the other two birds during all post-surgery measurement days. Furthermore, the individual with the highest CK 2 days after surgery had the greatest decline in descent speed for days 1–4 and 107 (excluding bird A for day 107). Causes of myopathy in birds include nutritional deficiencies, capture and handling, extreme exertion, and some medications (Aktas et al. 1997; Guglielmo et al. 2001; Shivaprasad et al. 2002; Abbott et al. 2005). We suggest that exertion (during pen capture, application of anesthesia, and post-operative handling), surgical trauma (including incisions), and possibly hyperthermia (maximum temperatures during surgery were 40.2–42.1° C) may have contributed to elevations in CK. While it appears there may be a relationship between slower descents, loss of power per stroke, and CK we remain cautious; CK has not been experimentally evaluated as a myopathy index for Common Eiders and we cannot isolate the cause of elevated blood CK. It is possible the surgical process alone could cause most or all of this elevation. More study is needed before this relationship can be fully evaluated.

2.5.2 *CONCLUSIONS*

Although it is premature to conclude that the effects reported here would alter health or fitness of birds in the wild or the validity of PTT-derived data (survival, movement, etc.), it is possible that all could be affected given the importance of dive performance to eiders. Based on the outcome of our study, further research on the effects of implanted PTTs and the development of improved implant procedures and transmitter design may be warranted. To better understand how implanted transmitters affect sea ducks, we recommend the following: (a) empirically test how energetics are affected, (b) address how PTT mass may influence effects, (c) differentiate effects at different stages of the PTT implant process (i.e., capture, surgical, and post-operative), (d) determine if other implanted devices without percutaneous antennas, such as time depth recorders (TDRs), have similar effects, (e) establish if effects are constant across taxa, (f) investigate if effects can be reduced by marking birds during specific seasons, and (g) assess if and when dive performance returns to baselines. Additionally, based on dive performance changes reported here and their possible effects to foraging ecology, we advise caution when combining implanted transmitters with percutaneous antennas and dive information devices such as TDRs.

Findings that Common Eiders' dive speeds are altered for months after transmitter implantation are significant. Previous studies on the effects of implanted transmitters in sea ducks generally only reported elevated short-term mortality (e.g., Iverson et al. 2006) and that mass, return rates, and most biomarkers were unchanged

1-yr after implantation of a transmitter with percutaneous antenna (Esler et al. 2000; Mulcahy et al. 2007). Researchers should consider our results and those of other applicable studies when establishing appropriate data censor periods and determining the efficacy of implanted PTTs in sea ducks for particular questions.

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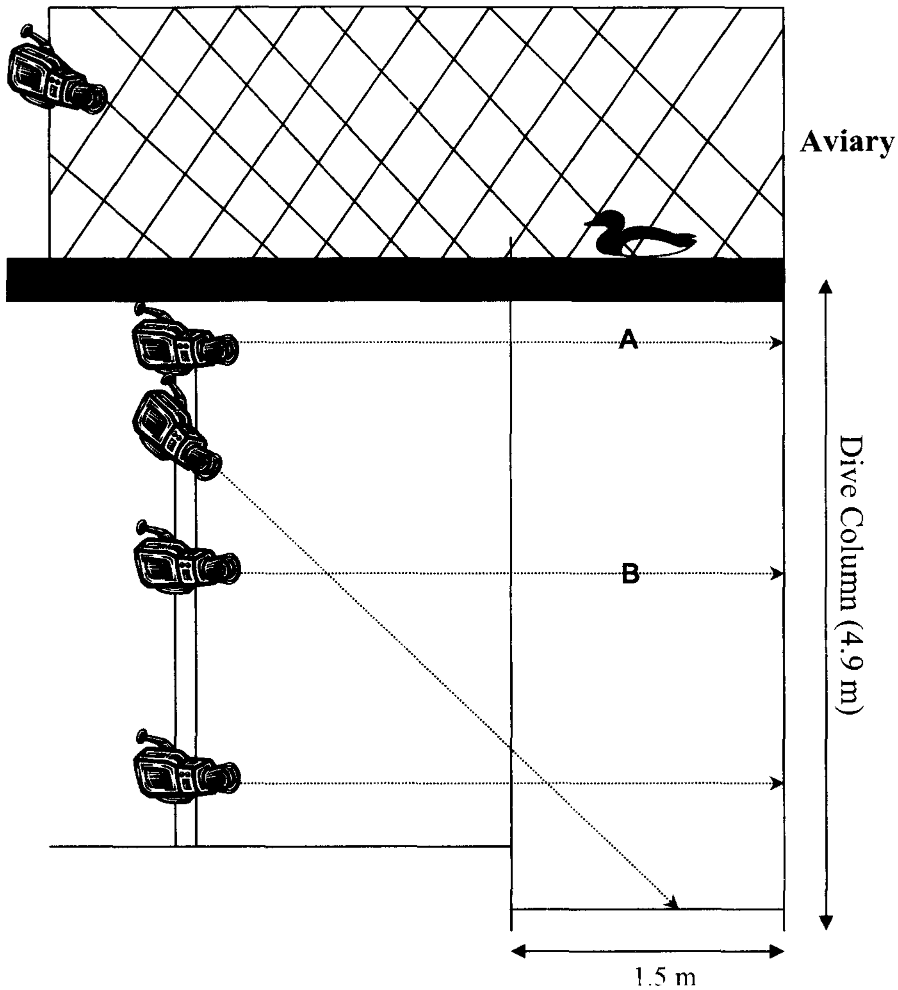

Figure 2.1 Illustration of dive column and surrounding aviary. Three cameras captured diving, one camera captured behavior and foot strokes on bottom, and 1-3 cameras captured surface activity.

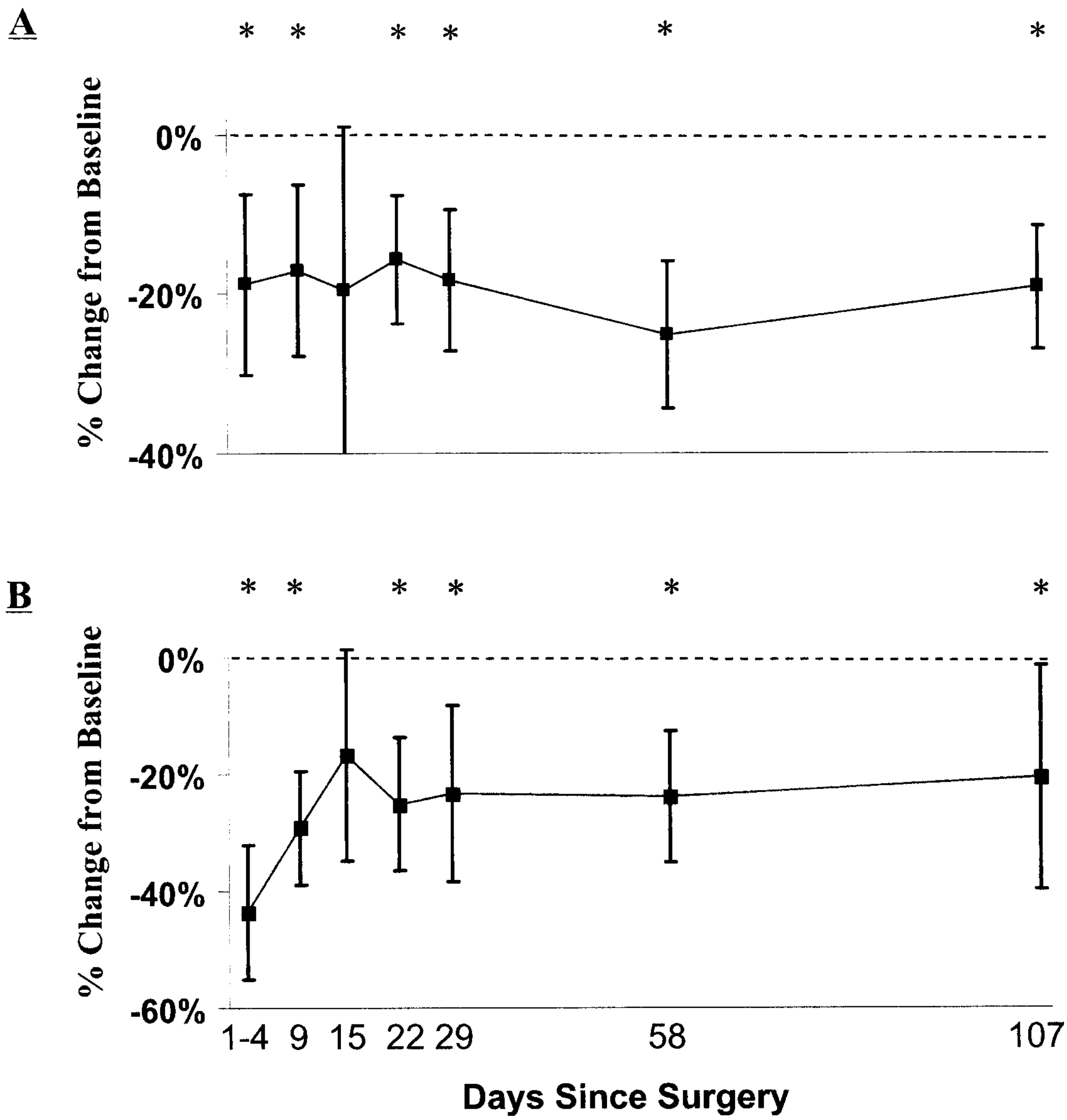

Figure 2.2 Mean % change in median descent and ascent speeds. Average percent change ± SD of median (A) descent and (B) ascent rates of six Common Eiders implanted with 38-47 g satellite transmitters with percutaneous antennas. * indicates significant difference between baseline (horizontal dashed line) and post-implant day with 1-tailed paired *t*-test after applying Holm-Bonferroni procedure.

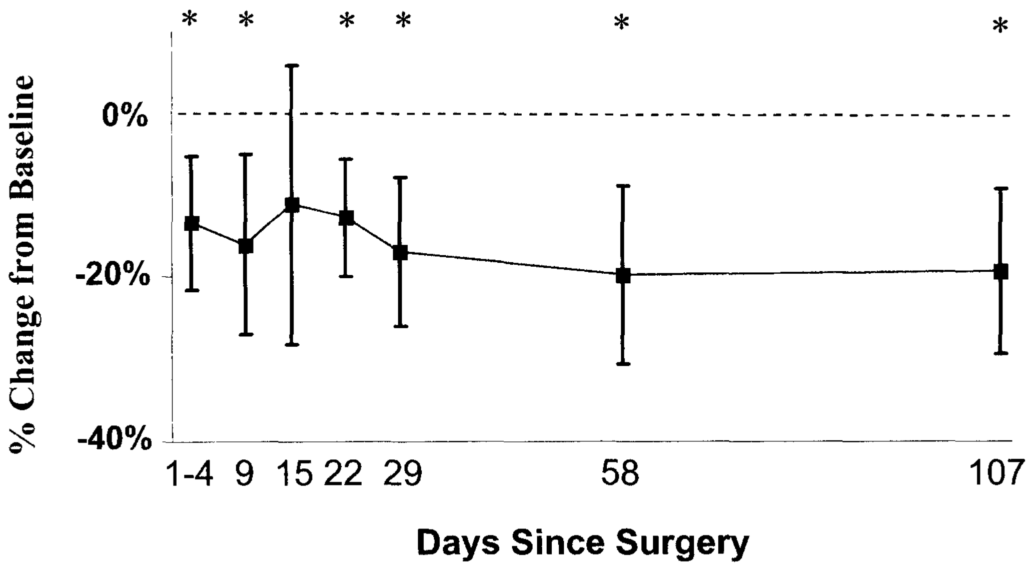

Figure 2.3 Mean % change in median descent speeds (coordinated dives only).

Average percent change ± SD of median coordinated propulsion descent rates (excluding dives in which birds used propulsion other than coordinated foot/wing strokes) of six Common Eiders implanted with 38-47 g satellite transmitters with percutaneous antennas. * indicates significant difference between baseline (horizontal dashed line) and post-implant day with 2-tailed paired *t*-test.

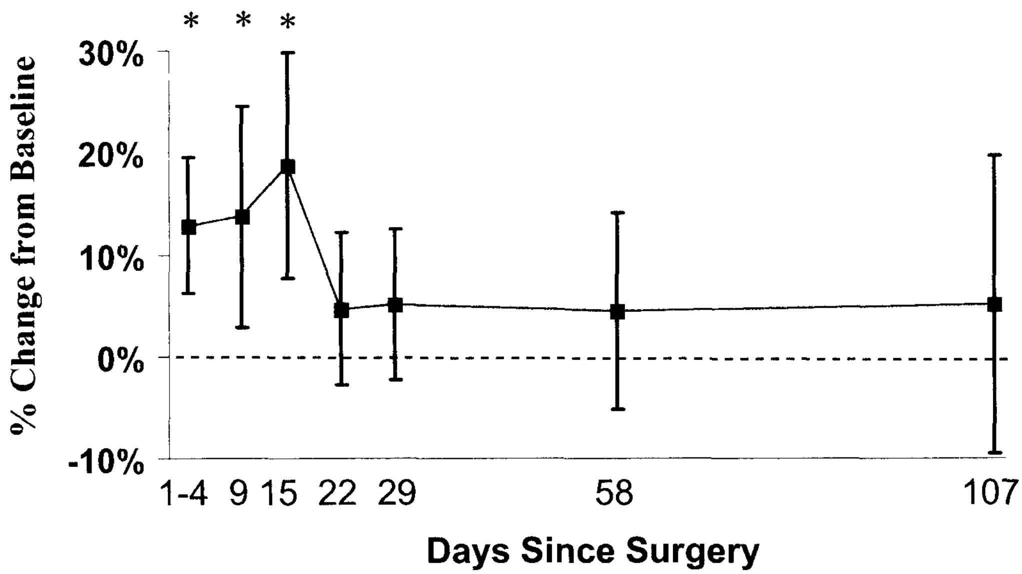

Figure 2.4 Mean % change in median dive duration. Average percent change ± SD of median dive duration of foraging dives of six Common Eiders implanted with 38-47 g satellite transmitters with percutaneous antennas. * indicates significant difference between baseline (horizontal dashed line) and post-implant day with 2-tailed paired *t*-test.

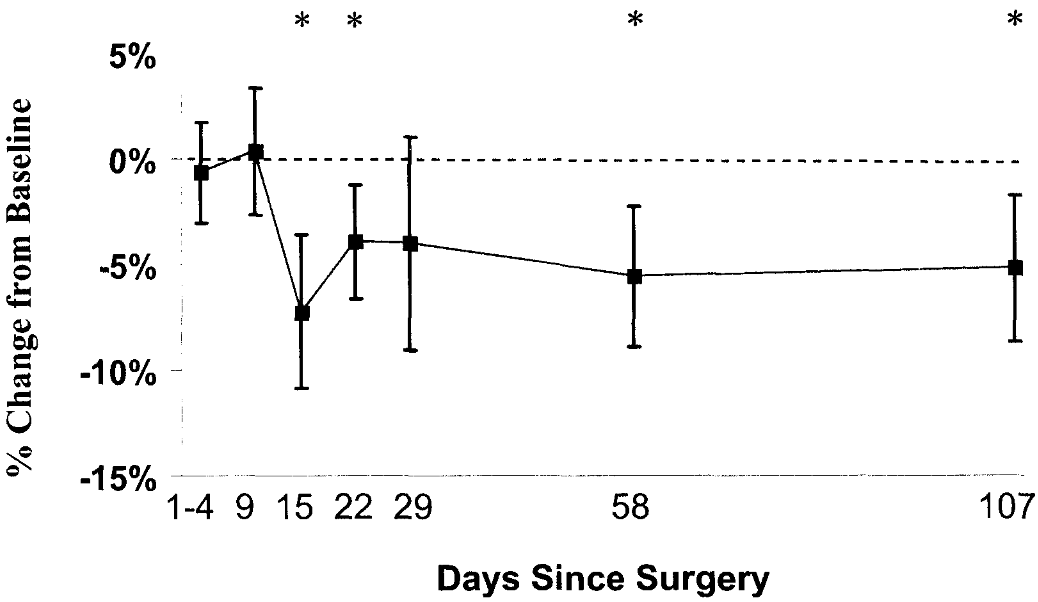

Figure 2.5 Mean % change in foot stroke frequency while foraging on bottom. Average percent change ± SD of Common Eiders' foot stroke frequency while foraging on the bottom after abdominal implantation of 38-47 g satellite transmitters with percutaneous antennas (n = 6). * indicates significant difference between baseline (horizontal dashed line) and post-implant day with 2-tailed paired *t*-test.

Table 2.1 Summary of primary dive parameters. Primary dive parameter values from six Common Eiders surgically implanted with 38–47 g satellite transmitters with percutaneous antennas. We recorded birds diving within a 4.9 m deep dive column and determined baselines (2–4 days pre-surgery) and post-operative values (1–107 days). Results of paired *t*-tests are presented, with *P* values from one-tailed analyses for descent and ascent rates and two-tailed analyses for stroke frequencies.

Parameter	Days Since Implant	Average Median ± SD	*t*	*P*
Descent Rate ($m\ s^{-1}$)	Baseline	0.99 ± 0.06		
	1–4	0.80 ± 0.14	4.2	0.004*
	9	0.82 ± 0.13	3.9	0.006*
	14–15	0.80 ± 0.23	2.3	0.03
	22	0.83 ± 0.11	5.2	0.002*
	29	0.81 ± 0.12	5.2	0.002*
	58	0.74 ± 0.11	6.7	<0.001*
	107	0.80 ± 0.09	6.0	<0.001*
Ascent Rate ($m\ s^{-1}$)	Baseline	1.09 ± 0.23		
	1–4	0.60 ± 0.07	7.0	<0.001*
	9	0.76 ± 0.10	6.2	<0.001*
	14–15	0.89 ± 0.21	2.2	0.04
	22	0.80 ± 0.11	5.2	0.002*
	29	0.81 ± 0.05	3.7	0.007*
	58	0.81 ± 0.09	4.8	0.003*
	107	0.84 ± 0.11	2.7	0.02
Descent Stroke Frequency (strokes s^{-1})	Baseline	2.36 ± 0.09		
	1–4	2.28 ± 0.10	2.4	0.06[a]
	9	2.24 ± 0.12	3.1	0.03[a]
	14–15	2.25 ± 0.11	2.1	0.10[a]
	22	2.29 ± 0.07	2.0	0.10[a]
	29	2.28 ± 0.13	1.3	0.28[a]
	58	2.20 ± 0.10	3.1	0.03[a]
	107	2.25 ± 0.09	2.9	0.03[a]

[a] Overall F-test non-significant. Paired tests should be viewed as exploratory.
[*] Indicates statistical significance after adjustment with Holm-Bonferroni.

Table 2.2 Summary of exploratory dive parameters. Exploratory dive parameter values of six Common Eiders surgically implanted with 38–47 g satellite transmitters with percutaneous antennas. We recorded birds diving within a 4.9 m deep dive column and determined baselines (2–4 days pre-surgery) and post-operative values (1–107 days). Statistics are provided from paired *t*-tests. *P* values for coordinated foot/wing descent rate are one-tailed and others two-tailed.

Parameter	Days Since Implant	Average median values ± SD	*t*	*P*
Coordinated Foot/Wing Descent Rate ($m\ s^{-1}$)[a]	Baseline	1.01 ± 0.06		
	1–4	0.88 ± 0.11	4.0	0.005
	9	0.85 ± 0.11	3.5	0.009
	14–15	0.90 ± 0.19	1.6	0.09
	22	0.88 ± 0.08	4.0	0.005
	29	0.84 ± 0.11	4.5	0.003
	58	0.81 ± 0.11	4.2	0.004
	107	0.81 ± 0.10	4.4	0.004
Dive Duration (s)	Baseline	28 ± 2		
	1–4	32 ± 4	-4.9	0.005
	9	32 ± 3	-3.3	0.02
	14–15	33 ± 3	-4.6	0.006
	22	30 ± 4	-1.5	0.19
	29	30 ± 2	-1.7	0.15
	58	29 ± 3	-1.0	0.35
	107	30 ± 3	-0.7	0.50
Bottom Foot Stroke Frequency (strokes s^{-1})	Baseline	3.22 ± 0.13		
	1–4	3.20 ± 0.15	0.7	0.52
	9	3.23 ± 0.11	-0.2	0.82
	14–15	2.99 ± 0.15	4.9	0.005
	22	3.10 ± 0.12	3.6	0.02
	29	3.09 ± 0.10	2.0	0.11
	58	3.05 ± 0.22	4.2	0.008
	107	3.06 ± 0.16	3.6	0.02

[a] Coordinated foot/wing stroke descent rates exclude dives where birds used forms of propulsion other than coordinated foot/wing strokes.

Table 2.3 Summary of primary dive parameters excluding bird with foot necrosis. Primary dive parameter values of Common Eiders implanted with 38–47 g PTTs with percutaneous antennas excluding bird A (male that developed necrosis of a toe which required partial amputation) (n = 5). We recorded birds diving within a 4.9 m deep dive column and determined baselines (2–4 days pre-surgery) and post-operative values (1–107 days). Results of paired *t*-tests are presented, with *P* values from one-tailed analyses for descent and ascent rates and two-tailed analyses for stroke frequencies.

Parameter	Days Since Implant	Average Median ± SD	*t*	*P*
Descent Rate ($m\ s^{-1}$)	Baseline	0.98 ± 0.06		
	1–4	0.80 ± 0.15	3.4	0.01
	9	0.83 ± 0.14	3.1	0.02
	14–15	0.81 ± 0.25	1.8	0.08
	22	0.84 ± 0.12	4.3	0.007
	29	0.81 ± 0.13	4.2	0.07
	58	0.76 ± 0.11	6.3	0.002
	107	0.82 ± 0.08	8.2	<0.001
Ascent Rate ($m\ s^{-1}$)	Baseline	1.16 ± 0.18		
	1–4	0.61 ± 0.06	7.3	0.001
	9	0.78 ± 0.09	7.6	<0.001
	14–15	0.92 ± 0.22	2.3	0.04
	22	0.82 ± 0.11	11.0	<0.001
	29	0.82 ± 0.05	6.2	0.002
	58	0.83 ± 0.07	6.2	0.002
	107	0.84 ± 0.12	4.4	0.006
Descent Foot/Wing Stroke Frequency (strokes s^{-1})	Baseline	2.35 ± 0.10		
	1–4	2.27 ± 0.11	1.9	0.12
	9	2.25 ± 0.13	2.5	0.07
	14–15	2.25 ± 0.12	1.5	0.20
	22	2.30 ± 0.07	1.4	0.22
	29	2.29 ± 0.14	0.8	0.45
	58	2.21 ± 0.11	2.4	0.07
	107	2.23 ± 0.09	2.7	0.05

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General Conclusions:

This study is the first to document the physiological responses of Common Eiders to implanted PTTs. Few studies have measured responses to implanted transmitters in sea ducks, and most have concentrated on ecological parameters such as survival. I used physiological and behavioral metrics to gain insight into how birds cope with surgery and carrying of a transmitter. Changes I found suggest birds in the wild could be affected by reduced foraging efficiency and increased energy demands for weeks to months after surgery. When combined with clinical findings, such changes might help to explain short-term mortality observed in the wild in some sea duck PTT studies, and add support for data censor periods.

In Chapter 1, I found that all primary biomarkers and six of nine secondary markers changed in response to PTT implantation. In period A (2–14 days post-implant), creatine kinase (CK), aspartate aminotransferase (AST), heterophil:lymphocyte ratio (H:L), and β_1-, β_2-, and γ-globulins were elevated from baselines; and albumin:globulin ratio (A:G), packed cell volume (PCV), and albumin were decreased from baselines. Additionally, I noted shivering on the day of surgery, lethargy on the first few days after surgery, breaks in waterproofing until day 14, feather matting at antenna exit sites until day 21, and preening at antenna exit sites after surgery. Despite the observed biochemical and clinical responses, mass increased overall between days 1 and 14 post-surgery. In period B (21– 28 days post-implant), AST, fecal glucocorticoid metabolites (FGMs), H:L, β_1-, β_2-, and γ-globulins, and PCV remained deviated from baselines, although the degree of change was generally less than observed in period A. Period C (2–3.5 months post-implant) had the fewest changes; only β_1- and γ-globulins and PCV were deviated from baselines and in each case, the degree of change was less than earlier periods.

In Chapter 2, I found both descent and ascent speeds for 4.9 m dives were slower for 3.5 and 2 months, respectively, after surgery. Additionally, I found dive duration was longer for 2 weeks post-surgery, and foot stroke frequency while birds foraged on the bottom was slower for most days between 14 and 107 days post-surgery. Despite an approximate 20% decrease in descent speed, I found no change in foot/wing stroke frequency during descent implying power per stroke was lower after surgery.

Results suggest that Common Eiders implanted with PTTs experienced myopathy, increased physiological stress, higher energetic requirements, lower aerobic dive limits (based on lower PCV, proposed myopathy, and higher underwater travel costs), and reduced diving efficiency. Also, lower PCV and albumin could be viewed as indicative of poorer general health. Most biomarkers returned to baseline levels during the post-surgery study period (3.5 months). Only two of nine secondary markers (β1- and γ-globulins) and PCV were different from baselines beyond 1 month post-surgery, and the extent of change for all three was less than earlier periods. One of the most interesting findings was that despite biomarkers generally normalizing within a month of surgery, dive performance remained affected at the end of the project. Although descent was the only primary dive parameter statistically different 107 days after surgery, ascent remained on average 21% slower with five of the six birds slower than baseline on this day. Additionally, when I removed one bird with foot necrosis from my analyses, ascent rate for the remaining birds was significantly slower than baseline for all dates, including day 107. This is one example of how biologically important changes may be missed by using a strictly statistical approach, especially with small sample sizes and variation in individuals' responses. From over a decade of studies using implanted PTTs in sea ducks, we know that effects of these devices do not generally lead to mortality, but that elevated mortality sometimes does occur within the first few weeks of surgery. Based on this observation we do not expect all birds to respond the same clinically, biochemically, or behaviorally. When sample sizes are large, statistical tests probably provide the best insight into central tendencies, but if samples are small, large changes in some birds may lead to statistical insignificance despite most or all birds having values deviated from baselines.

In Chapter 1, I lumped data into time periods to minimize the number of tests, utilize all three baseline values, and account for variation in how individuals respond temporally to stimuli. While pooling data and taking maximum (minimum) values during the different periods accomplished these tasks, it also limited my ability to track patterns among an individual's chemistries. For example, interpreting muscle enzymes in relation to one another on specific collection dates is often beneficial and allows comparisons based on pharmacokinetic differences between the enzymes. The lumping of data in Chapter 1 made such contrasts more difficult. Had I provided readers with plots of each individual, time point, and chemistry (essentially graphs of the raw data), such a comparison would have been easier and, in some cases, may have provided a more thorough assessment. Such an approach might also have led to additional conclusions at the individual level; the bird with a very high CK value on day 2 (F3) also had high corresponding LDH values, despite LDH not being statistically affected during this period. This shows that although birds did not universally have elevated LDH, some birds had changes that were individually important. Additionally, because the birds acted as their own controls, I could not be certain that PTT implantation was the only important variable in this study. Although I attempted to address this issue by using extended baselines from the year prior to surgery (Chapter 1), the lack of true control limits the inferences I can make.

While I found many changes in response to implanted transmitters, the ultimate effect on eiders in the wild remains uncertain. Some differences between captive birds and wild birds were clear, though the causes of these differences were not. Birds in this study dove and ascended slower than wild Common Eiders. This could have occurred because of the dive column itself (circular ascent commonly seen post-surgery was probably due, in part, to limited space), dive depth, differences among sub-species, and/or physical fitness of the captive birds. For example, had birds here had unlimited space, spiraling during ascent may not have occurred; however, the angle of ascent may have remained affected, thus ascent remaining slower.

In addition, our captive eiders only performed a few dozen foraging dives each day, compared to the hundreds performed daily by wild eiders (Guillemette 2001). If the captive birds had to perform more dives, fly, search for prey, and be exposed to stochastic at-sea conditions, the extent of change in health and stress chemistries may have been greater. In contrast, for some changes such as descent speed, lack of physical activity on par with wild birds may have caused the degree of change in captive birds to be greater than had birds been in peak physical condition. With respect to changes in mass, effects of *ad libitum* feeding has been shown in other studies. For example, captive Mallards with external transmitters released at two habitat types (a natural tidal marsh and a private shooting area where birds were provided supplemental grain) exhibited different responses in mass post-release. Mallards with access only to environmental foods lost an average of 10.2 g per day, whereas birds with access to supplemental food gained an average of 7.7 g per day (Rohwer et al. 2002). Furthermore, Harlequin Ducks implanted with transmitters lost more weight than did banded controls during the first few weeks after surgery (Esler et al. 2000). Thus, while a captive study allows inference to wild birds, care is needed in assessing ultimate effects on eiders under natural pressures.

While the aforementioned limitations constrained my ability to assess if and how PTT-measured parameters might be affected, some concern for data bias is warranted. For example, changes in travel times could have important consequences to wild eiders. In Chapter 2, I extrapolated to show that for deeper dives (11.3 m), implanted Common Eiders might have 14-21% less time to aerobically forage than unmarked birds. Wild Common Eiders could compensate for reduced dive times in several ways, including increasing the number of dives performed or changing foraging behavior. If we consider a theoretical situation where wild Common Eiders implanted with PTTs concentrated their foraging efforts in shallower areas than unimplanted conspecifics, habitat data derived from PTTs would be biased. While this scenario relies on supposition, it seems almost certain that if wild eiders do experience changes in travel times at or near those presented here, some repercussions are inevitable.

This study was a step forward in elucidating responses of sea ducks to implanted transmitters. Understanding these responses is crucial for deciding device efficacy and interpreting the data these devices provide. Further research is needed to confirm findings (using a larger sample size and control subjects), define causality of changes, and experimentally determine if energetics are affected. Research should also assess if changes in transmitter design or the implant procedure might minimize effects.

Estimating what changes in transmitter design or the transmitter implantation would reduce effects is difficult because I did not evaluate definitive causes of most responses. That said, minimizing the density and volume of implanted PTTs should reduce effects on balance and buoyancy and therefore could reduce changes in ascent rate and behavior. I found some indications that myopathy may play a role in slower descent speeds, but more research is necessary to confirm this and determine why elevations of muscle enzymes occurred. It is important to note that the term myopathy is defined as a disorder of muscle tissue and is not specific to a particular cause and does not define the biological importance or length of such a disorder. While captive birds habituated to handling should not be affected by capture myopathy in the traditional sense, captive birds still struggle during pre- and post-surgery handling and the application of anesthesia that may lead to some degree of exertional myopathy. Other causes could include hyperthermia, surgical incisions, or bruising during the implant procedure. Future research using control groups that are handled the same as implanted birds, as well as a sham surgery group, might reveal why muscle enzymes in the blood were elevated. Pre-surgical intervention may also be of benefit. Wild Bobwhite Quail (*Colinus virginianus*) captured in walk-in traps and marked with external radiotransmitters pre-treated for myopathy with vitamin E and selenium had higher survival than did birds without pretreatment (Abbott et al. 2005).

The effects of biochemical responses might be mitigated to some degree if birds are implanted during periods without additional energetic needs (e.g., during harsh weather, periods of reduced food availability, and during molt). Conditions such as plumage wetting may have a greater effect on birds during cold inclement weather. Overall, effects might be minimized by implanting birds on the breeding grounds where food is often available at shallow depths, cover provides birds with some protection from avian predators, and environmental conditions are not typically as harsh as those experienced at sea.

Ultimately, the need for information on particular ecological questions may override concern for effects to birds or derived data. In some situations, bias introduced by PTT-effects may not substantially limit the usefulness of data. When the value of data would be affected, researchers should use data censor periods or an alternate method of data collection. Censor period length should be based on known effects of implanted transmitters for specific species, and the particular questions researchers wish to use PTTs to address. While a two-week censor period may be appropriate for assessing survival for some sea duck species (Esler et al. 2000, Iverson et al. 2006), based on changes in dive performance reported here, a longer censor period may be needed when addressing differential habitat use, especially if depth varies between locations. Abdominally implanted PTTs with percutaneous antennas are and will remain useful in assessing important ecological questions about migration, movement patterns, and habitat use for sea ducks. Researchers planning studies using these devices should be mindful of effects reported here and results from other studies when deciding device suitability and determining adequate censor periods to ensure the validity of data for extrapolation to unmarked conspecifics.

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Appendix. Summary of biomarker responses for all post-surgery dates. Biomarker responses of common eiders implanted with 38-47 g satellite transmitters with percutaneous antennas. Blood collected on the day of surgery was collected prior to the first bird being moved from the dive column to pre-op and ~ 0.5-3.0 hrs prior to the first incision. n = 6 unless noted.

Parameter	Collection Date	Mean ± SD	Parameter	Collection Date	Mean ± SD	Parameter	Collection Date	Mean ± SD
CK	441 d pre-surgery	178 ± 123	**A:G**	441 d pre-surgery	1.01 ± 0.08	**PCV**	441 d pre-surgery	43 ± 3.1
(U/L)	363 d pre-surgery	141 ± 56	(ratio)	363 d pre-surgery	1.10 ± 0.08	(% cells)	363 d pre-surgery	51 ± 4.2
	323 d pre-surgery	174 ± 99		323 d pre-surgery	1.06 ± 0.13 [a]		323 d pre-surgery	52 ± 3.1
	258 d pre-surgery	157 ± 56 [a]		258 d pre-surgery	0.89 ± 0.14 [a]		258 d pre-surgery	49 ± 3.6 [a]
	211 d pre-surgery	174 ± 52		211 d pre-surgery	0.92 ± 0.04		211 d pre-surgery	49 ± 2.9
	62 d pre-surgery	358 ± 364		62 d pre-surgery	1.09 ± 0.10		62 d pre-surgery	48 ± 1.9
	29 d pre-surgery	345 ± 261		29 d pre-surgery	1.02 ± 0.10		29 d pre-surgery	51 ± 4.6
	day of surgery	234 ± 107		day of surgery	1.00 ± 0.12		day of surgery	50 ± 2.4
	2 d post-surgery	3049 ± 4667		2 d post-surgery	0.66 ± 0.08		2 d post-surgery	41 ± 3.1
	8 d post-surgery	137 ± 59		8 d post-surgery	0.61 ± 0.10		8 d post-surgery	43 ± 2.8
	14 d post-surgery	141 ± 77		14 d post-surgery	0.75 ± 0.12		14 d post-surgery	45 ± 2.6
	21 d post-surgery	509 ± 533		21 d post-surgery	0.89 ± 0.12		21 d post-surgery	45 ± 2.3
	28 d post-surgery	312 ± 275		28 d post-surgery	0.89 ± 0.13		28 d post-surgery	47 ± 4.0
	56 d post-surgery	475 ± 478		56 d post-surgery	0.93 ± 0.07		56 d post-surgery	47 ± 3.3
	91 d post-surgery	154 ± 44		91 d post-surgery	1.07 ± 0.11		91 d post-surgery	48 ± 3.7
	105 d post-surgery	186 ± 103		105 d post-surgery	0.92 ± 0.17		105 d post-surgery	48 ± 1.8

Appendix cont.

FGM (ng/g)	3 d pre-surgery	11.0 ± 4.8
	2 d pre-surgery [c]	9.3 ± 9.2 [c]
	1 d pre-surgery	10.7 ± 5.7
	3 d post-surgery	16.7 ± 2.9
	6 d post-surgery	13.3 ± 7.1
	13 d post-surgery	18.0 ± 7.6 [a]
	20 d post-surgery	24.6 ± 19.3
	27 d post-surgery	23.4 ± 14.7
	55 d post-surgery	15.7 ± 7.0
	104 d post-surgery	14.7 ± 4.8

LDH (U/L)	62 d pre-surgery
	29 d pre-surgery
	day of surgery
	2 d post-surgery
	8 d post-surgery
	14 d post-surgery
	21 d post-surgery
	28 d post-surgery
	56 d post-surgery
	91 d post-surgery
	105 d post-surgery

AST (U/L)	62 d pre-surgery	0.5 ± 1.2
	29 d pre-surgery	0 ± 0
	day of surgery	1.2 ± 2.9
	2 d post-surgery	17.5 ±19.5
	8 d post-surgery	8.8 ±14.7
	14 d post-surgery	10.3 ±12.8
	21 d post-surgery	26.3 ±19.3
	28 d post-surgery	0 ±0
	56 d post-surgery	0 ± 0
	91 d post-surgery	0 ± 0
	105 d post-surgery	0 ± 0

α1-globulins (g/dL)	62 d pre-surgery
	29 d pre-surgery
	day of surgery
	2 d post-surgery
	8 d post-surgery
	14 d post-surgery
	21 d post-surgery
	28 d post-surgery
	56 d post-surgery
	91 d post-surgery
	105 d post-surgery

768 ± 182	β1-globulins (g/dL)	62 d pre-surgery	0.25 ± 0.03
722 ± 509		29 d pre-surgery	0.25 ± 0.03
568 ± 245		day of surgery	0.25 ± 0.03 [b]
1356 ± 1517		2 d post-surgery	0.25 ± 0.03
546 ± 107		8 d post-surgery	0.38 ± 0.05 [a]
463 ± 144		14 d post-surgery	0.32 ± 0.02
558 ± 129		21 d post-surgery	0.30 ± 0.04
507 ± 106		28 d post-surgery	0.27 ± 0.04
693 ± 160		56 d post-surgery	0.30 ± 0.03
595 ± 182		91 d post-surgery	0.24 ± 0.03
590 ± 223		105 d post-surgery	0.29 ± 0.06
0.88 ± 0.09	β2-globulins (g/dL)	62 d pre-surgery	0.55 ± 0.05
1.06 ± 0.04		29 d pre-surgery	0.45 ± 0.08
0.98 ± 0.04 [b]		day of surgery	0.54 ± 0.24 [b]
0.87 ± 0.12		2 d post-surgery	0.80 ± 0.30
1.06 ± 0.07 [a]		8 d post-surgery	0.98 ± 0.23 [a]
1.01 ± 0.09		14 d post-surgery	0.98 ± 0.25
0.99 ± 0.08		21 d post-surgery	0.84 ± 0.18
0.97 ± 0.09		28 d post-surgery	0.87 ± 0.14
1.05 ± 0.06		56 d post-surgery	0.67 ± 0.12
0.87 ± 0.12		91 d post-surgery	0.59 ± 0.10
1.07 ± 0.11		105 d post-surgery	0.72 ± 0.34

Appendix cont.

γ-globulins (g/dL)	62 d pre-surgery	0.16 ± 0.02	**Glucose** (mg/dL)	62 d pre-surgery
	29 d pre-surgery	0.15 ± 0.03		29 d pre-surgery
	day of surgery	0.14 ± 0.04 [b]		day of surgery
	2 d post-surgery	0.12 ± 0.03		2 d post-surgery
	8 d post-surgery	0.19 ± 0.06 [a]		8 d post-surgery
	14 d post-surgery	0.23 ± 0.08		14 d post-surgery
	21 d post-surgery	0.24 ± 0.08		21 d post-surgery
	28 d post-surgery	0.21 ± 0.08		28 d post-surgery
	56 d post-surgery	0.17 ± 0.08		56 d post-surgery
	91 d post-surgery	0.19 ± 0.02		91 d post-surgery
	105 d post-surgery	0.17 ± 0.04		105 d post-surgery
Albumin (g/dL)	62 d pre-surgery	2.01 ± 0.19	**H:L** (ratio)	62 d pre-surgery
	29 d pre-surgery	1.95 ± 0.24		29 d pre-surgery
	day of surgery	1.79 ± 0.19 [b]		day of surgery
	2 d post-surgery	1.34 ± 0.21		2 d post-surgery
	8 d post-surgery	1.60 ± 0.17 [a]		8 d post-surgery
	14 d post-surgery	1.88 ± 0.12		14 d post-surgery
	21 d post-surgery	2.10 ± 0.28		21 d post-surgery
	28 d post-surgery	2.06 ± 0.29		28 d post-surgery
	56 d post-surgery	2.04 ± 0.24		56 d post-surgery
	91 d post-surgery	2.03 ± 0.25		91 d post-surgery
	105 d post-surgery	2.01 ± 0.21		105 d post-surgery

[a] n = 5
[b] n = 4
[c] n = 2
[d] test not performed for this date

218 ± 29
214 ± 22
215 ± 21
204 ± 17
194 ± 22
216 ± 10
218 ± 11
207 ± 18
209 ± 24
[d]
207 ± 18
0.32 ± 0.17
0.58 ± 0.40 [b]
1.04 ± 0.75
3.15 ± 2.09
1.47 ± 0.80
1.31 ± 0.86
1.55 ± 0.66
0.91 ± 0.30
0.97 ± 0.71
0.58 ± 0.15
0.67 ± 0.42